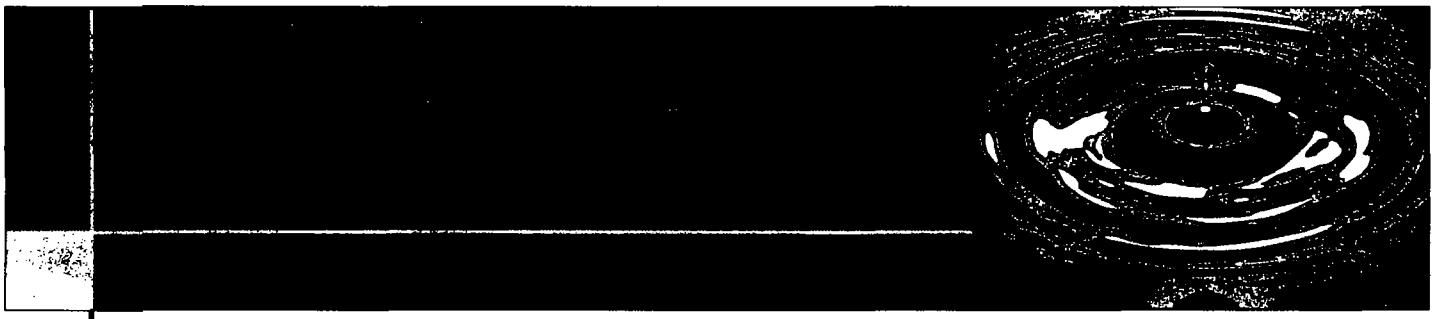

FIELD SAMPLING PLAN (FSP)
POTOSI SITE - WASHINGTON COUNTY LEAD DISTRICT
WASHINGTON COUNTY, MISSOURI
MAY 2008

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Acronyms and Abbreviations

AE	Assessment Endpoints
AUF	Area Use Factor
BERA	Baseline Ecological Risk Assessment
$CaCO_3$	Calcium Carbonate
CLP	Contract Laboratory Program
COC	Chain of Custody
COPC	Chemical of Potential Concern
DMF	Dominant Macroinvertebrate Families
DO	Dissolved Oxygen
Eco-SSL	Ecological Soil Screening Level
EISOPQAM	Environmental Investigation Standard Operating Procedures and Quality Assurance Manual
ERA	Ecological Risk Assessment
ERAGS	Ecological Risk Assessment Guidelines for Superfund
FSP	Field Sampling Plan
HASP	Health and Safety Plan
IDW	Investigation Derived Waste
MDC	Missouri Department of Conservation
MDNR	Missouri Department of Natural Resources
MSCI	Macroinvertebrate Stream Condition Index
MS/MD	Matrix spike/matrix spike duplicate
NVSS	Non-Volatile Suspended Solids
ORP	Oxidation Reduction Potential
PPM	Parts per Million
PO	Potosi
QA	Quality Assurance
QA/QC	Quality Control/Quality Assurance
QAPP	Quality Assurance Project Plan
RI/FS	Remedial Investigation/Feasibility Study
SEM/AVS	Simultaneous extractable metals/acid volatile sulfides
SHAD	Stream Habitat Assessment Device
SLERA	Screening Level Ecological Risk Assessment
SOP	Standard Operating Procedure
TAL	Target Analyte List
TOC	Total Organic Carbon
USEPA or EPA	United States Environmental Protection Agency
WCLD	Washington County Lead District

TABLE OF CONTENTS

1.0. INTRODUCTION

An Ecological Risk Assessment (ERA) is an evaluation of the potential impacts of contaminants of potential concern on ecological receptors. As part of the focused Remedial Investigation/Feasibility Study (RI/FS), an ERA must be performed to identify unacceptable risks to the environment and evaluate the effectiveness of various remedial alternatives to address such risks. This Field Sampling Plan (FSP) describes the elements to be included in a Baseline Ecological Risk Assessment (BERA), which will be conducted according to the Ecological Risk Assessment Guidance for Superfund (EPA, 1997). The objective of this BERA, in particular, will be to characterize risk to the biological function of aquatic and terrestrial ecosystems exposed to metal contamination resulting from historical mining activities in the Potosi Site in Washington County, Missouri.

2.0. SITE BACKGROUND

The Washington County Lead District Site (WCLD) is part of Missouri's Old Lead Belt, which is located in the Ozark Mountains in southeast Missouri (Figure 1). The Old Lead Belt provided approximately 80 percent of the lead produced in the United States. In Washington County, the first mines were mostly surface diggings advanced by manual labor with a pick and shovel. Starting in 1799, deeper mines were started in the area. Additionally, this area is part of the barite mineralization district of Missouri. After the Civil War, numerous small barite mines operated in Washington County in the early 1900s. Barite mining boomed in 1926 as the mineral's use for oil drilling mud was discovered and for a number of years, Washington County was the world's leading producer of barite before declining in the 1980s. Many of the later large mining operations reworked lands that were previously hand mined for galena (mineral source of lead) or barite. Washington County has hosted over 1,000 lead and barite mining, milling, or smelting sites.

The WCLD site actually contains three separate NPL sites, the Richwoods site, the Old Mines site, and the Potosi site (Figure 2). Each site contains soil, ground water, surface water and sediment contaminated with arsenic, barium, cadmium, and lead associated with historical mining practices in southeast Missouri. The Potosi site encompasses approximately 40 square miles in the central eastern portion of Washington County, Missouri and contains mined areas and tailings ponds in nine Study Areas (SA-1 through SA-9) (Figure 2).

3.0. HABITAT AND ECOLOGY

The existing information describing the habitat and ecology of the area includes maps, aerial photography, and several ecological studies. *A Biological Assessment Report for Mill Creek* (MDNR, 2006) describes the condition of the main tributary on the site. This study evaluated the habitat and aquatic macroinvertebrate communities of this important stream. Also, the Missouri Department of Conservation's (MDC) Big River Inventory and Assessment, which includes fish, mussel, crayfish, aquatic insect and habitat data for the streams and tributaries found within the site, also contains important information pertaining to the ecology of the site. Several EPA Region 7 Conservation Opportunity Area maps were also referred to in an effort to identify high priority terrestrial and aquatic conservation areas that may occur within the site boundaries (MORAP, 2005). Finally, general references, such as *The Fishes of Missouri* (Pflieger, 1997) and *The Wild Mammals of Missouri* (Schwartz *et al.*, 2001) were consulted.

3.1. Surface Water and Sediment

The Potosi site is drained by Mill Creek and its associated tributaries (Figure 3). Mill Creek flows northeasterly from Potosi, Missouri to its confluence with the Big River near Blackwell, Missouri. The classified section of the stream is approximately 12 miles long and is categorized as a Class "P" stream (MDNR, 2005), which means that it maintains flow during periods of drought. The stream has beneficial use designations for livestock and wildlife watering, protection of warm water aquatic life, human health fish consumption and whole body contact (MDNR, 2005).

The Mill Creek watershed is known to be affected by mine related activity. Shibboleth Branch, a downstream tributary to Mill Creek, was placed on the 2002 303(d) list of impaired waters in Missouri for Non-Volatile Suspended Solids (NVSS) or sediment (MDNR, 2002; EPA 2007). A barite tailing dam breach in 1975 on Shibboleth Branch impacted Mill Creek and Big River with sediment for approximately 9 months. Fountain Farm Branch may contribute barite mining sediment to Mill Creek (MDNR, 1994). Pond Creek, an upstream tributary to Mill Creek, is 303 (d) listed for sediment from barite mining sources (MDNR, 2002). In 2002-2003, MDNR identified elevated dissolved barite levels in the Big River with probable origins in the Mill Creek watershed (MDNR, 2004)

Water chemistry samples collected as part of MDNR's Biological Assessments showed that dissolved metal concentrations in Mill Creek did not exceed state or federal chronic water quality criteria. However, dissolved barium concentrations do exceed Great Lakes Water Quality Initiative Tier II values (secondary chronic values), which provides an ecological screening level for barium given the lack of state or federal water quality criteria.

To date, sediment chemistry samples from the site have not been collected and analyzed for metal contamination.

3.2. Fish

Several locations in the Mill Creek watershed were sampled using seining and electrofishing techniques as part of the MDC's Biotic Inventory and Assessment for the Big River watershed (Figure 4). Fish species lists from each sampling location within theWCLD, as well as total abundance, are provided in Table 1 (Appendix A). Some of the most frequently collected fishes were typical Ozark species, such as smallmouth bass (*Micropterus dolomieui*), rock bass (*Amploplites rupestris*), longear sunfish (*Lepomis megalotis*), northern hogsucker (*Hypentelium nigricans*), black redhorse (*Moxostoma duquesnei*), greenside darter (*Etheostoma blennioides*), rainbow darter (*Etheostoma caeruleum*), Ozark minnow (*Notropis nubilus*), and striped shiner (*Luxilus chrysocephalus*). Rainbow trout have been stocked in Mill Creek by a private business in Cadet, Missouri, but the numbers and stocking frequency are unknown.

Four species of concern are found within the basin, but only the Silverjaw Minnow (*Ericymba buccata*) has been sampled within the WCLD. The crystal darter (*Ammocrypta asprella*) is on the State endangered list and was found in lower Big River (River Mile (RM) 1-8), but only by Pflieger (1975). The Missouri status of the Alabama shad (*Alos alabamae*) is rare and was infrequently sampled in Big River (RM 1-4). The western sand darter (*Ammocrypta clara*) is on Missouri's watch list and was found in lower Big River (RM 1-10). Silverjaw minnow (*Ericymba buccata*) distribution was scattered throughout Big River and lower portions of Calico Creek, Terre Bleue Creek, Flat River, and Salem Creek. It is on the State watch list, but was common where sampled.

To date, fish tissue samples from the Mill Creek watershed have not been collected and analyzed for metals.

3.3. Mussels

The Big River basin has a diverse mussel community. Thirty-four species of mussels have been found within basin streams (Buchanan 1980; Ryckman *et al.*, 1973) (Table 2, Appendix A). Three species are of special concern and were sampled only in lower Big River. The pink mucket (*Lampsilis abrupta*) is Federally-endangered, while the scaleshell (*Leptodea leptodon*) and spectaclecass (*Cumberlandia monodonta*) are listed as rare in Missouri. However, the most recent mussel survey conducted by Buchanan in 1980 did not include surveys

of Mill Creek and associated tributaries, therefore, it is unknown if any of these species occur within the site.

3.4. Crayfish

The Big River basin contains eight species of crayfish, including the belted crayfish (*Orconectes harrisoni*), which is found only in Missouri and almost exclusively in the St. Francois and Big River basins (one isolated Meramec River sample at the mouth of Big River). With the exceptions of the golden crayfish (*Orconectes luteus*) and the devil crayfish (*Cambarus diogenes*), all species are found only in Ozark streams. A complete species list for crayfish occurring in the Big River basin can be found in Table 3 (Appendix A).

Based on the results of the BERA for the Big River Mine Tailings Site, which is also located in the Old Lead Belt, crayfish appear to accumulate metals at a higher level compared with fish. To date, no crayfish tissue samples have been collected from the Mill Creek watershed and analyzed for metals.

3.5. Aquatic Insects

The Big River basin benthos communities can be quite diverse (Ryck, 1973). Twelve orders, 55 families, and 107 taxa of aquatic insects have been collected since 1976. Mayfly and stonefly nymphs were especially prevalent (Ryck 1973), generally indicating good water quality. However, in places, benthos populations are seriously affected by lead and barite mine waste (Ryck, 1973; Duchrow, 1976; Kramer, 1976). Duchrow (1976) found that invertebrates with exposed gills like dobsonfly (Nigronis serricornis) larvae may become extirpated if mine waste is present.

The *Biological Assessment Report* for Mill Creek (MDNR, 2006) included both habitat and biological assessments. The biological assessment was conducted using a Semi-quantitative Macroinvertebrate Stream Bioassessment Project Procedure. The data was analyzed using Macroinvertebrate Stream Condition Index (MSCI) scores, individual biological criteria metrics, and dominant macroinvertebrate families (DMF). The MSCI is a qualitative index of a stream's aquatic biological integrity. MSCI scores can range from non-supportive of the biological community to that of fully supporting the biological community (based on reference conditions). The results for Mill Creek showed that it is fully supporting of the biological community. Examination of the composition of dominant macroinvertebrate families and the individual taxa list identified a diverse and intolerant community. Overall, the macroinvertebrate community appears to be unimpaired. However, the assessments recommend further surveys be conducted on Mineral Fork tributaries Mine A Breton Creek, Clear Creek, and Old Mines Creek, as well as

further surveys on Mill Creek tributaries Fountain Farm Branch, Pond Creek, and Shibboleth Branch. The full Biological Assessment Report can be found in Appendix A.

3.6. Wildlife

The wildlife of Washington County is typical of the Ozarks. A species list for Washington County, provided by the Missouri Department of Conservation, can be found in Table 4 (Appendix A). Two federally listed bat species, the Gray Bat (*Myotis grisescens*) and the Indiana Bat (*Myotis sodalis*) occur in Washington County. Gray bats require undisturbed caves, and forage over streams, rivers and reservoirs. A corridor of mature trees between caves and foraging sites is important. Indiana bats hibernate in the winter in limestone caves. Summer habitat includes mature riparian and adjacent upland forests. Full forest canopy with open understory is preferred. Snags and cavity trees (greater than 9" dbh) are also important. Indiana bats forage in riparian forest and over water.

In addition, two state listed species occur in Washington County, the Northern Harrier Hawk (*Circus cyaneus*) and the Plains Spotted Skunk (*Spilogale putorius*). The Northern Harrier is a rare summer resident and uncommon winter resident of Missouri. It inhabits open fields, prairies, native grass plantings and shallow marshes. Northern Harriers require dense herbaceous vegetation, with nearly 100% canopy cover, reaching a height of 10" by mid-may. The Plains Spotted Skunk is very rare in Missouri. It inhabit fencerows, vegetated gullies and brushy borders with logs, brush piles, snags, rocky outcrops, open prairies, and riparian woodland areas.

Based on results from BERAs conducted at other mining sites in the region (EPA, 2006), the terrestrial exposure pathway that frequently drives these ecological risk assessments is the intake of soil by ground feeding insectivores, also known as vermivores. Vermivores are sensitive species for two reasons. First, there is a relatively higher percentage of soil (hence metals) in their diets. Second, the soil invertebrates they consume have a relatively higher metal concentration in their tissue. Generally, there are two species that represent vermivores well, and they are the short-tailed shrew (class mammalia) and the American woodcock (class aves). Both species can be found in Washington County.

3.7. Habitat

3.7.1. Aquatic Habitat: Stream habitat quality was evaluated using MDC's Stream Habitat Assessment Device (SHAD Version II) (Figure 5). SHAD was utilized at 113 sites on 49 streams. Overall, SHAD surveys revealed streambanks were in good condition in the Big

River Basin. The Big River and tributary streambanks showed minimal to no bank erosion. Trees and shrubs were the dominant types of streambank protection.

Based on the Shad results it is known that Shaw (2.0 miles), Shibboleth (0.5 miles), and Fountain Farm (0.2 miles) branches continue to be affected by barite mining sediment, which can smother aquatic habitats within these streams. Also, one instream sand and gravel mining operation is permitted on the Mineral Fork in Washington County, although the exact location is not known. These operations negatively affect adjacent stream reaches through increased channel instability (downcutting and channel widening), streambank erosion, increased turbidity, and loss of aquatic habitat. In other areas, watershed urbanization has decreased wooded riparian corridors and increased stormwater runoff, thereby increasing channel instability.

Riparian corridor condition was fair to poor. Forty-four percent of the tributaries exhibited a timbered stream corridor > 75 ft, but timbered corridor was absent all together on another forty four percent of tributaries. Twenty-one percent of tributary SHAD sites had corridors that consisted mainly of grasses. Cattle grazing and hay production are prevalent land uses around these tributaries, and corridor width is being reduced along streams with increasing amounts of urbanization.

Results from SHAD surveys suggest that the potential for soil erosion and non-point source pollution may be greater from tributary streams than from Big River, due to heavier riparian corridor land use and poor vegetative quality (narrow corridor and prevalence of grasses). Cattle's grazing increases erosion and greatly limits the development of wooded corridors. Row cropping and hay production eliminates wooded corridors through constant plowing or mowing. Generally, intensive riparian corridor land use and poor vegetative quality increased as the size of stream decreased.

Big River basin's instream habitat is typical of Ozark streams with gravel present at 89% of the SHAD sites. Water willow bordering pools and boulder slides from bluffs was common. Seventy-four percent of the tributary streams had downed logs or rootwads.

Nineteen sensitive natural communities have been documented within the basin (MDC, 1995). Included in these communities are two examples of Ozark creeks and four examples of Ozark springs and spring branches.

3.7.2. Terrestrial Habitat: The WCLD is located in the Ozark Highland eco-region and is situated within the Salem Plateau section of the Ozark physiographic province (Fenneman, 1938). The Site topography can be described as hilly and is located within a region of several hundred feet of relief and altitudes ranging from 700 to 1,000 feet above mean sea level (msl. The Ozark highlands are dominated by oak and oak-hickory forests.

3.8. Soil

Barite and Lead (galena) mineralization in the sample area occurs in fractured and solution bedrock and red clay residuum derived chiefly from the Cambrian Potosi and Eminance dolomite. Some mineralization in veins and small isolated residual deposits is associated with the Ordovician Dolomite in northeastern Washington County and southeastern Franklin County. The majority of the barite mines were processed from residuum. The residuum is often capped at the surface by layers of barren soil that ranges from several inches to three to four feet deep. In many areas, vegetation now covers the previously mined areas, with mine waste intermingling with the soil.

3.9. Vegetation

The vegetation present at the Site is similar in composition to that typical of the region. The climax vegetation community in the Ozarks is mature oak-hickory forest. Due to over 300 years of human activity, other vegetation communities at the Site are also important. Such communities include; urban areas, pasture, cultivated farmlands, constructed wetlands, second growth oak and hickory, and several stands of short leaf pine. To date, floral community surveys or investigations of metal concentrations in vegetation have not been conducted.

3.10. Soil Invertebrates

Insects, including soil invertebrates, known to occur in Washington County, can be found in the species list in Table 4. Previous soil invertebrate chemistry investigations and toxicity testing have not been conducted.

4.0. PROJECT SCOPE AND INVESTIGATION

Additional information is needed to evaluate the exposure and effects of the chemicals of potential concern (COPC) to ecological assessment endpoints (AE). This FSP completes Step 4 of the Ecological Risk Assessment Guidelines for Superfund (ERAGS). The purpose of this ecological field sampling event is to collect representative data necessary to support the completion of the Baseline Ecological Risk Assessment (BERA) as part of the RI/FS process. The data gaps include the following:

- Soil –Soil sampling throughout the site is needed to characterize both impacted and background (reference) conditions.

- Surface Water – Although some data exists for surface water conditions in Mill Creek, the data is not comprehensive enough to fully characterize the watershed. Additional data including filtered samples as well as hardness and pH measurements are needed to fill this gap. The focus of surface water characterization efforts should be on the identification of a diverse set of stream segments and waterbodies that are representative of both impacted and background (reference) conditions.

- Sediment Sampling – Sediment data is needed to fully characterize the watershed. Future sediment samples should be co-located with the surface water samples and come from a diverse set of stream segments and waterbodies that are representative of both impacted and background (reference) conditions.

- Stream Habitat Evaluation – Habitat in the stream reaches should be evaluated to supplement the macroinvertebrate surveys that will be conducted, as well as to provide critical information to be used in targeting sites for potential remediation.

- Macroinvertebrate Community Survey – Existing information indicates that the macroinvertbrate communities of the Mill Creek watershed are relatively healthy. However, additional community surveys will be performed in locations throughout the watershed, including tributaries that are known to have been directly impacted by mining.

- Sediment Toxicity Testing – As an additional line of evidence, sediment toxicity testing will be done at the stream locations in which the macroinvertebrate surveys were conducted.

- Terrestrial Biota Sampling – To address potential risk to sensitive terrestrial species that consume earthworms and other soil invertebrates, earthworm sampling, co-located with soil samples should be conducted to characterize varied conditions from different habitats and soil conditions. In addition, vegetation samples should be collected to model risks to herbivores and omnivores as well as to address risk to plant communities.

- Aquatic Biota Sampling – Aquatic biota samples (including crayfish, and small forage fish) should be collected to fully determine COPC concentrations and the potential risks to organisms that feed on them.

- Crayfish – No crayfish data is available. Crayfish sampling is therefore needed to characterize COPC concentrations in aquatic invertebrates and potential risks to organisms that feed on them.

- Small Fish – Fish data is needed to fully characterize the impacts to fish communities as well as the organisms that feed on them.

5.0. FIELD ACTIVITIES

The ecological field sampling activities will include several investigations that will fill certain data gaps and provide data to support the BERA as part of the RI/FS process. The technical approach, including the type and numbers of environmental samples to be collected, the methods of chemical analyses, and specific sampling locations, are subject to change pending further discussions between the EPA and other stakeholders.

5.1. Sampling Procedures

The ecological field sampling activities will provide data necessary to complete the BERA as part of the RI/FS process. It will include the following tasks:

- Site Reconnaissance
- Surface Soil Investigation
- Surface Water and Sediment Investigation
- Biota Investigation (terrestrial and aquatic).

5.1.1. Site Reconnaissance: The purpose of site reconnaissance is to determine field conditions prior to the start of field work to enable field activities to start on time and within schedule and budget. In addition, this site reconnaissance meets the criteria for Step 5 of the ERAGS and will enable EPA to determine if the target species and samples can actually be collected during the site visit. This activity will be conducted in July, 2008 and includes a site visit by the ecological risk assessment team for the purpose of determining sample access conditions and staking out sample locations. Access letters will also be distributed to nearby residents whose properties would need to be accessed for the ecological field sampling event. During site reconnaissance activities, the sample stations will be located and verified using a Trimble Pro XRS Global Positioning System (GPS). This system uses satellite correction to enable sub-meter horizontal accuracy.

5.1.2. Surface Soil Investigation: The surface soil investigation will include the collection of 20 surface soil samples (17 samples, 2 background samples, and 1 duplicate) from mining impacted areas from the 0- to 12-inch depth interval. The background locations were chosen at locations believed to be unimpacted by mining activities based on preliminary site investigation data. Because the metal contamination in the region is so widespread, EPA will use a background lead concentration of 60 mg/kg in soil as a reference. All sampling will be performed following the EPA-ERT Standard Operating Procedures (SOPs). The surface soil sampling SOP is included in Appendix B. The proposed surface soil sample locations are identified on Figure 6. The sample designations, descriptions, and rationale are listed in Table 5. Surface soils will be collected from the following locations:

Study Area-1: SS17
Study Area-2: SS16, SS15, SS18 (background)
Study Area-3: SS13, SS14
Study Area-4: SS12, SS12-FD
Study Area-5: SS19 (background), SS11, SS10
Study Area-6: SS09, SS08
Study Area-7: SS07
Study Area-8: SS05, SS04, SS03, SS06
Study Area-9: SS02 , SS01

All surface soil samples (including background) will be analyzed under the EPA Contract Laboratory Program (CLP) for the following constituents: Target Analyte List (TAL) total metals with the following physical characteristics: percent moisture, total organic carbon (TOC), and field pH. Sample collection containers, requirements, preservation, and holding times are presented in Table 7 (Appendix A) for all samples.

5.1.3. Surface Water and Sediment Investigation: The surface water and sediment investigation will include the collection of thirty-six sediment and surface water samples. There will be 34 sediment/surface water locations, plus one background location, and one duplicate location. The sediment and surface water sample locations are co-located with each other to determine the nature and extent of contamination and to fill data gaps from existing data sets. Each sediment sample will be collected from the 0 to 6-inch depth interval using the appropriate grab sampling technique. In tailings ponds, bulk sediment will be collected. In streams, the sediment will be sieved to capture the fine grained material. Based on the chemistry results for the fine grained material, a gradient of metal contamination will be established for use in a 10-day *Hyallela azteca* toxicity test.

Each surface water sample will be collected from the 0 to 12-inch depth interval using the grab technique and filtered in the field. All sampling and toxicity testing will be performed according to the EPA-ERT Standard Operating Procedures (SOPs) which are included in Appendix B. Sediment and surface water sample locations are presented in Figure 7. The sample designations, descriptions, and rationale are listed in Table 7 (Appendix A). Sediment and surface water will be collected from the following locations:

Study Area-1: SD33-SD34, SW33-SW34
Study Area-2: SD28-SD32, SW28-SD32, SD35 (background), SW35 (background)
Study Area-3: SD24-SD27, SW24-SW27
Study Area-4: SD21-SD23, SW21-SW23
Study Area-5: SD20, SD20-FD, SW20, SW20-FD
Study Area-6: SD15-SD19, SW15-SW-19
Study Area-7: SD11-SD14, SW11-SW114
Study Area-8: SD06-SD10, SW06-SW-10
Study Area-9: SD01-SD05, SW01-SW05

EPA will also verify in the field that the background sample locations are taken at elevations greater than the local natural outcropping of lead. All sediment samples will be analyzed under the CLP for TAL metals, TOC, and percent moisture.

All surface water samples will be analyzed under the CLP for TAL total metals, dissolved metals and hardness. Field parameters including pH, temperature, specific conductance, turbidity, dissolved oxygen (DO), and Oxidation Reduction Potential (ORP) using a Horiba water quality meter will also be recorded at each location.

5.1.4. Biota Investigation: The biota investigation will include the collection of earthworm and vegetation samples from terrestrial areas and crayfish and small forage fish samples from aquatic areas. All biota samples will be collected in accordance with the EPA-ERT SOPs presented in Appendix B.

5.1.4.1. Earthworm and Vegetation Tissue Sampling. Earthworm and vegetation tissue samples will be collected at a subset of 10 of the soil sampling locations (co-located locations). The earthworm samples from each location will be composited in the laboratory and one sample per location will be analyzed for TAL total metals and lipid content. In the event that earthworms are not found at the specified locations, an alternative soil sampling location will be identified to ensure the collection of all ten samples. Similarly, the vegetation samples from each location will be composited in the laboratory and one sample per location will be analyzed for TAL total metals

5.1.4.2. Crayfish Tissue Sampling. Crayfish are an important food source in the aquatic food chain and will be collected to better characterize COPC concentrations in aquatic invertebrates and to evaluate their potential risk to organisms that feed on them. Ten crayfish samples will be collected from Mill Creek and associated tributaries. The samples will be collected from the same locations as the sediment and surface water samples. The samples will be composited in the laboratory into one sample per location for whole body analysis.

5.1.4.3. Small Fish Tissue Sampling. Fish samples will be collected from a variety of tailings ponds and stream segments. Ten small fish samples will be co-located with the crayfish samples (discussed above in Section 5.1.4.2.). The targeted small fish species will include small species that feed on other fish and benthic invertebrates, and will be captured via electroshocking and nets. Fish collection procedures are discussed in the SOP for biota sampling presented in Appendix B. The samples will be composited in the laboratory into one sample per location for whole body and fillet analysis. The fish samples will be analyzed for TAL metals and lipid content.

5.1.4.4. Macroinvertebrate Community Survey. Macroinvertebrate communities will be surveyed in sections of Mill Creek, Mine A Breton Creek, Pond Creek, and Bates Creek using a Kick net. The SOP for macroinvertebrate sampling can be found in Appendix B. Samples will be collected and brought back to the laboratory for identification. The following metrics will be calculated: total counts, taxa richness, EPT Index, % EPT, Ratio EPT divided by Chironomidae abundance, % dominance, % Chironomidae, and dominant taxa.

5.2. Quality Assurance/Quality Control Sampling

As part of the sampling effort, quality assurance/quality control (QA/QC) samples will be submitted to the laboratory with field investigation samples in order to evaluate the confirmatory sampling procedures and analytical methodologies. The types of QA/QC samples that will be collected and their corresponding frequencies include the following:

- Matrix spike/matrix spike duplicate (MS/MSD) – 1 in 20 samples collected per media
- Field blank – 1 per week
- Rinsate equipment blank – 1 per week
- Duplicates – 5% of total samples collected per media

As part of the sampling program, QA/QC samples will be submitted to the laboratory along with the corresponding field investigative samples.

6.0. SAMPLE DESIGNATIONS

A sample numbering system will be used to identify each sample for analysis. The purpose of this numbering system is to provide a tracking system for retrieval of data on each sample. All sample numbers will begin with the Potosi site location code (PO); they will be designated with a Study Area (SA) number. There are 9 SAs within the site (Figure 2).

Proposed sample designations, descriptions, locations, and rationale are presented in Table 5.

6.1. Surface Soil Samples

All surface soil samples will be designated with the PO location code, followed by the Study Area number, followed by "SS" and a pre-determined surface soil station location. A typical surface soil sample collected from SA-1 would be designated as PO-01-SS17.

6.2. Surface Water Samples

All surface water samples collected from tailings ponds and streams will be designated with the PO location code, followed by the Study Area number, followed by "SW" and a pre-determined surface water station location. A typical surface water sample collected from a tailing pond in SA-09 would be designated as PO-09-SW01.

6.3. Sediment Samples

All sediment samples collected from tailings ponds and streams will be designated with the PO location code, followed by the Study Area number, followed by "SD" and a pre-determined sediment station location. The stream samples will also be followed with an "S" to indicate that the sediment has been sieved. A typical sediment sample collected from a tailings pond in SA-2 would be designated as PO-02-SD19. A typical sediment sample collected from Mill Creek in SA-9 would be designated PO-09-SD01-S.

6.4. Biota Samples

All biota samples will be designated with the PO location code, followed by the Study Area number, followed by one of the following biota type acronyms and a pre-determined sediment station location. The biota type acronyms include the following:

- EW – earthworms
- CR – crayfish
- SF – small fish
- VG - Vegetation

A typical earthworm sample collected from Study Area 9 would be designated as PO-09-EW01, a crayfish sample collected from Study Area 1 would be designated as PO-01-CR01; and a small fish sample collected in a tailing pond from Study Area 8 would be designated as PO-08-SF01.

6.5. QA/QC Samples

6.5.1. Matrix spike/matrix spike duplicates. Samples designated for use as an MS/MSD will retain their original sample designation but will be identified on the sample label and traffic report/chain of custody record (TR/COC) as a "sample used for QC".

6.5.2. Field Blanks. All field blanks will be designated with "FB" followed by the sample designation from the location in which they were collected and date marker in the "MMDDYY" format. For example, a field blank collected at surface soil location SS01 in SA-1 would be designated as: WCLD_FB_SA-1_SS01_MMDDYY.

6.5.3. Rinsate Blanks. All rinsate blanks will be designated with "RB" followed by the sample designation from the field activity in which they were collected and date marker in the "MMDDYY" format. For example, a rinsate blank collected after decontamination at a sediment location would be designated as follows: WCLD_RB_MC_SD01_MMDDYY.

6.5.4. Duplicate Samples. All field duplicate samples will retain the same sample designation as the corresponding field sample, but will be identified on the label and TR/COC record with a "FD" placed at the end of the station identifier number.

7.0. INVESTIGATION PROCEDURES AND METHODS

This section describes the field procedures and methods to be utilized throughout this field investigation. Specific field procedures and methods have been selected for use to ensure that sampling and data collection activities are conducted within acceptable QA standards. Additional information concerning QA protocols is presented in the Quality Assurance Project Plan (QAPP).

7.1. Summary of Sampling Program

All surface soil, surface water, sediment, and biota samples will be collected from locations as described in Section 5.0. All samples will be analyzed by EPA accepted methods as presented in the QAPP.

The objective of the sampling and preservation procedures outlined in this FSP and QAPP is to obtain samples that yield consistently high quality data. The use of proper sampling equipment, strict controls in the field, and appropriate chain-of-custody and analytical procedures will reduce the potential for sample misrepresentation and unreliable analytical data.

Specific sampling and preservation procedures to be used in the field investigation are detailed in the following sections. Standard operating procedures (SOPs) for all operations are included in Appendix B of this FSP. The EPA Region VII Environmental Investigation Standard Operating Procedures and Quality Assurance Manual (EISOPQAM) was the primary document from which all field procedures were developed (EPA, 1997). Container, preservation, and holding time requirements must also meet the requirements of the EISOPQAM (EPA, 1997). Table 7 presents the container, preservation, and holding time requirements for this project. The EPA's CLP Guidance for Field Samplers will be the guidance used for performing CLP sampling techniques. All personnel conducting sampling will be experienced in implementing the sampling procedures as outlined herein.

7.1.1. Sampling Equipment and Procedures. All field sampling will be conducted in accordance with the SOPs included in Appendix B. These SOPs have been developed to optimize sample integrity and representativeness. The SOPs contain specifications for the types of sampling equipment that will be used and the procedures that will be followed during sample collection activities. The collection of site samples will also be guided by procedures developed to provide the appropriate level of worker protection as detailed in the site- and task-specific Health and Safety Plans (HASPs), and by quality assurance and quality control procedures as detailed in the QAPP. The sample collection activities and corresponding SOPs that will be utilized during this field sampling activities for site are listed in the table below.

8.0. CONTAMINATION PROCEDURES

Specific procedures for equipment decontamination will be implemented to avoid cross-contamination of surface and subsurface strata and samples of various media, which are to be submitted for chemical analyses. Decontamination procedures will meet or exceed the requirements of the EISOPQAM (EPA, 1997). All sampling equipment will be decontaminated between sample locations at each specific sampling location. The following procedures will be used for all sampling equipment:

1. Clean with tap water and Alconox phosphate-free, low-foaming, neutral soap using a brush.

If it is necessary to remove particulate matter and surface films the following procedures are performed:

1. Rinse thoroughly with tap water.
2. Rinse thoroughly with analyte free water.
3. Rinse with 0.1 molar (M) nitric acid.
4. Rinse thoroughly with organic/analyte free water.
5. Remove the equipment from the decontamination area and cover with plastic or aluminum foil.

Equipment stored overnight will be covered and sealed with clean, unused plastic or aluminum foil. Each time the sampling equipment is decontaminated, it should be treated as if it were the final decontamination so that no contaminants are transported offsite or to other sampling locations.

9.0. SAMPLE HANDLING AND ANALYSIS

9.1. Sample Containment and Preservation

Sample containment and preservation are as important to any environmental sampling event as the procedures by which the samples are collected. Container requirements along with preservation procedures and holding times are presented in Table 7 of the FSP.

9.2. Sample Collection Documentation

Sample collection documentation procedures are another vital aspect of any environmental sampling event. Each sample or field measurement will be properly documented to facilitate timely, correct, and complete analysis.

9.2.1 Field Operation Records. The most important aspect of sample collection documentation is thorough, accurate record keeping. The documentation of field operations associated with sample collection will be recorded on field sampling logs and in field logbooks. All field sampling logs are presented in Appendix B, with their respective SOP.

Region 7 field sheets will also be used to track access to properties, sample collection, and numbering from the chain of custody documents. The field team will complete a field sheet for each location sampled. Bound field notebooks will be maintained by the field team leader to provide daily records of significant events, observations, and measurements during the sampling events. Each page will be numbered, signed, and dated. The field team leader will maintain the field notebooks. These notebooks will be kept as permanent records. Field notebooks are intended to provide sufficient data and observations to enable participants to reconstruct events that occurred during projects and to refresh the memory of the field personnel if called upon to give testimony during legal proceedings. In a legal proceeding, notes, if referred to, are subject to cross-examination and are admissible as evidence. The field notebook entries should be legible, factual, detailed, and objective.

All original data recorded in field notebooks will be written in waterproof ink. These accountable, serialized documents are not to be destroyed or thrown away, even if they are illegible or contain inaccuracies that require a replacement document. If an error is made on an accountable document assigned to one person, that individual may make corrections simply by crossing out the error and entering the correct information. Any erroneous information should not be destroyed. The person who made the entry should correct any error discovered on an accountable document. All corrections must be initialed and dated.

Sample collection or other documentation may also take the form of photographs, which will be taken with either a digital camera or disposable camera and included in the permanent project file. The photographs may show the surrounding area and reference objects that identify the sampling locations. The digital camera will be programmed so that the date imprint appears on all photos. All photographs will be saved to a CD as part of the permanent project file. An entry will be made in the field log identifying which sampling location is depicted in each photograph. Logbook entries of photographs will have the following major components: photographer's initials roll, frame number, date, and a description of what was photographed.

9.2.2. Sample Custody Documentation. The sample chain-of-custody (COC) procedure provides another means of sample collection documentation. The sample chain-of-custody procedure documents the identifying, tracking, and monitoring of each sample from the point of collection through final data reporting.

10.0. INVESTIGATION-DERIVED WASTES

Various types of investigation-derived wastes (IDW) are defined in the EISOPQAM, Chapter 5 (EPA, 1997). Types of IDW anticipated to be generated at the Washington County Lead District Site include:

- Decontamination fluids
- Soil and sediment samples
- Uncontaminated wastes.

These types of IDW are anticipated to be generated as a result of the field sampling activities. Therefore, the following procedures and safeguards will serve as the pollution control and mitigation plan for the site.

- Decontamination Fluids. These fluids include wash waters used to decontaminate the sampling equipment. The wash waters will be disposed of at each specific sampling location either on to the ground or into the river/stream/pond.
- Uncontaminated Wastes. Packaging, trash, flagging, etc., will be placed in trash bags and disposed of in a dumpster at the R7 Science and Technology Center.

11.0. REFERENCES

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APPENDIX A
Tables

Table 1: Fish species Lists for Mill Creek and Mineral Fork and associated tributaries (MDC, 2003)

Stream	Site	Date	Species	# EFed	# seined	Total
Mineral Fork	Gay	09/17/2003 00:00	chestnut lamprey	1		1
			bigeye chub	2		2
watershed area:	76476 ac		bleeding shiner	165	84	249
order:	5		hornyhead chub	15	1	16
river mile:	12.9		ozark minnow	8	1	9
Sampled by:	A. Austin, D. Brown, S. Oakes		stoneroller	236	7	243
			striped shiner	12	1	13
EF Gear:	Barge		wedgespot shiner	4	11	15
EF Time:	1916		northern hogsucker	8		8
Siene time:	3 min. 10 sec.		slender madtom	2		2
			yellow bullhead	1		1
			blackspotted topminnow	1		1
			northern studfish	1	5	6
			mottled sculpin	42	1	43
			bluegill	34		34
			green sunfish	6		6
			hybrid sunfish	1		1
			largemouth bass	6		6
			longear sunfish	70		70
			rock bass	42		42
			smallmouth bass	4		4
			logperch	8		8
			greenside darter	12		12
			Missouri saddled darter		1	1
			orangethroat darter	1		1
			rainbow darter	7	2	9
			Totals	689	114	803

Stream	Site	Date	Species	# EFed	# seined	Total
Mineral Fork	Taylor	09/16/2003 00:00	bleeding shiner	245	113	358
			hornyhead chub	12		12
watershed area:	77142 ac		ozark minnow	23	8	31
order:	5		stoneroller	88	2	90
river mile:	12.2		striped shiner	7	1	8
Sampled by:	A. Austin, S. Oakes		wedgespot shiner	21	3	24
			black redhorse	3		3
EF Gear:	Barge		golden redhorse	1		1
EF Time:	2468		northern hogsucker	12		12
Siene time:	02:37		flathead catfish	1		1
			yellow bullhead	1		1
			blackspotted topminnow	3		3
			blackstripe topminnow		1	1
			northern studfish	2		2
			mottled sculpin	9	3	12
			bluegill	23		23
			green sunfish	4		4
			largemouth bass	12		12
			longear sunfish	51		51
			rock bass	21		21
			smallmouth bass	4		4
			logperch	5		5
			fantail darter		1	1
			greenside darter	6		6
			rainbow darter	2	1	3
			Totals	556	133	689

Stream	Site	Date	Species	# EFed	# seined	Total
Shibboleth Br.	Upstream from Tiff	07/21/2003 00:00	rosyface shiner		1	1
			redfin shiner		11	11
watershed area:	4999 ac		bleeding shiner	49	16	65
order:	3		bluntnose minnow	5	1	6
river mile:	1		creek chub	1		1
Sampled by:	A. Austin, D. Brown, S. Oakes		hornyhead chub	4		4
			ozark minnow	6	5	11
EF Gear:	Backpack		stoneroller	27		27
EF Time:	2280 sec.		striped shiner	1	3	4
Siene time:	153 sec.		golden redhorse	1		1
			northern hogsucker	1		1
			slender madtom	3		3
			black bullhead	1		1
			blackspotted topminnow	2	4	6
			blackstripe topminnow	1		1
			mottled sculpin	6		6
			bluegill	2		2
			green sunfish	5		5
			longear sunfish	15		15
			smallmouth bass	1		1
			johnny darter	1		1
			fantail darter	11	5	16
			greenside darter	10		10
			Missouri saddled darter	5		5
			orangethroat darter	1		1
			rainbow darter	7		7
			Totals	166	46	212

Stream	Site	Date	Species	# EFed	# seined	Total
Mill Creek	Govero (near Tiff)	07/23/2003 00:00	bigeye chub	2		2
			bleeding shiner	218	18	236
watershed area:	30585 ac		bluntnose minnow	14		14
order:	4		hornyhead chub	30		30
river mile:	2.9		ozark minnow	39		39
Sampled by:	Sarah Oakes, Andy Austin		rosyface shiner	4		4
			stoneroller	534	1	535
EF Gear:	Barge		striped shiner	4	3	7
EF Time:	1332		wedgespot shiner	2	1	3
Siene time:	3 min. 30 sec.		black redhorse	11		11
			golden redhorse	5		5
			northern hogsucker	12		12
			slender madtom	4		4
			blackspotted topminnow		1	1
			bluegill	1		1
			largemouth bass	2		2
			longear sunfish	22		22
			rock bass	40		40
			smallmouth bass	28		28
			spotted bass	5		5
			banded darter	1		1
			fantail darter	3		3
			greenside darter	20		20
			logperch	10		10
			Missouri saddled darter	11		11
			orangethroat darter	1		1
			rainbow darter	5		5
			Totals	1028	24	1052

Stream	Site	Date	Species	# EFed	# seined	Total
Mineral Fork	Rivermont Campground	07/24/2003 00:00	bigeye shiner		1	1
			bleeding shiner	164	97	261
watershed area:	101428 ac		hornyhead chub	10	1	11
order:	5		ozark minnow	8	7	15
river mile:	5.5		stoneroller	172	1	173
Sampled by:	Danny Brown, Andy Austin		striped shiner	10	3	13
			wedgespot shiner		2	2
EF Gear:	Tote Barge		golden redhorse	4		4
EF Time:	1347		northern hogsucker	6		6
Siene time:	4 min. 58 sec.		stone cat	1		1
			blackspotted topminnow	1		1
			blackstriped topminnow		1	1
			mottled sculpin	4		4
			bluegill	1		1
			largemouth bass	2		2
			longear sunfish	17		17
			rock bass	8		8
			smallmouth bass	4		4
			fantail darter	1		1
			greenside darter	5		5
			logperch	5		5
			Missouri saddled darter	15	1	16
			rainbow darter	6		6
			Totals	444	114	558

Stream	Site	Date	Species	# EFed	# seined	Total
Clear Creek	Last Chance Ranch	07/29/2003 00:00	bleeding shiner	29	6	35
			stoneroller	79		79
watershed area:	7624 ac		slender madtom	60		60
order:	3		blackspotted topminnow	2	10	12
river mile:	1.4		blackstripe topminnow	2	2	4
Sampled by:	Andy Austin, Sarah Oakes		northern studfish	12	8	20
			mottled sculpin	49		49
EF Gear:	Backpack		bluegill	2		2
EF Time:	1531		green sunfish	12		12
Siene time:	2 min. 10 sec.		longear sunfish	4		4
			smallmouth bass	1		1
			fantail darter	13	4	17
			orangethroat darter	4		4
			rainbow darter	13	1	14
			Totals	282	31	313

Stream	Site	Date	Species	# EFed	# seined	Total
Three Hill Creek	Co Rd 424	08/26/2003 00:00	bleeding shiner	47	14	61
			creek chub	8	1	9
watershed area:	4748 ac		ozark minnow	4		4
order:			southern redbelly dace	9		9
river mile:			stoneroller	86		86
Sampled by:	Sarah Oakes, Mike Reed		striped shiner	4	10	14
			northern hogsucker	5		5
EF Gear:	Backpack		slender madtom	18		18
EF Time:	1226		blackspotted topminnow		11	11
Siene time:	NA		blackstripe topminnow	7	15	22
			northern studfish	1		1
			mosquitofish		2	2
			mottled sculpin	4		4
			bluegill	3		3
			green sunfish	5		5
			hybrid sunfish	4		4
			longear sunfish	10		10

fantail darter	4		4
greenside darter	4	1	5
orangethroat darter	8		8
rainbow darter	4	1	5
Totals	235	55	290

Stream	Site	Date	Species	# EFed	# seined	Total
Mine a Breton	Wood's Farm	08/11/2003 00:00	bigeye chub	3	1	4
			bleeding shiner	194	21	215
watershed area:	27920 ac		creek chub	1		1
order:	4		hornyhead chub	26		26
river mile:	2.3		ozark minnow	14	2	16
			stoneroller	544	1	545
Sampled by : Andy Austin, Sarah Oakes			striped shiner	6		6
			black redhorse	36		36
EF Time (Barge) : 2221			golden redhorse	6		6
Seine time: 3 min. 17 sec.			northern hogsucker	12		12
			slender madtom	1		1
			yellow bullhead	10		10
			blackspotted topminnow	1		1
			blackstripe topminnow	2		2
			northern studfish	1	2	3
			mottled sculpin	43	8	51
			bluegill	17		17
			green sunfish	11		11
			hybrid sunfish	1		1
			largemouth bass	3		3
			longear sunfish	32		32
			rock bass	101		101
			smallmouth bass	41		41
			greenside darter	2	1	3
			rainbow darter	16	1	17
			Totals	1124	37	1161

Stream	Site	Date	Species	# EFed	# seined	Total
Allen Branch	Forche Valley Golf Course	07/30/2003 00:00	rainbow trout	1		1
			bigeye chub	246		246
watershed area:	4806.7 ac		bigeye shiner	4		4
order:	3		bleeding shiner	18	5	23
river mile:	0.4		creek chub	2		2
			golden shiner	2		2
Sampled by : Sarah Oakes, Andy Austin			ozark minnow	104	3	107
			stoneroller	59		59
EF Time (BP) : 1860			blackspotted topminnow	3	1	4
Seine Time : 2 min. 8 sec.			mottled sculpin	167	6	173
			green sunfish	7		7
			hybrid sunfish	1		1
			longear sunfish	12	1	13
			fantail darter	4		4
			greenside darter	26	2	28
			orangethroat darter	17	4	21
			rainbow darter	82	14	96
			Totals	755	36	791

Stream	Site	Date
Ebo Creek	Farrow Property	08/28/2003 00:00
watershed area:	3569 ac	
order:	3	
river mile:	0.8	
Sampled by: Danny Brown, Andy Austin		
EF Time (BP) : 1844		
Seine time : 3min 8sec		

Species	# EFed	# seined	Total
bleeding shiner	11	3	14
hornyhead chub	7	7	14
creek chub	14		14
southern redbelly dace	235		235
stoneroller	512	1	513
ozark minnow	2		2
slender madtom	28		28
blackspotted topminnow	6		6
blackstripe topminnow	4	7	11
northern studfish	4	1	5
mottled sculpin	73	4	77
green sunfish	13		13
hybrid sunfish	4		4
fantail darter	25	4	29
orangethroat darter	17		17
Totals	955	27	982

Table 2: Mussels found within the Big River watershed (Buchanan 1980, Ryckman et al., 1973).

Common Name	Scientific Name
Spectacle case	Cumberlandia monodonta
Paper floater	Anodonta imbecilus
Giant floater	Anodonta grandis grandis
Squaw foot	Strophitus undulatus undulatus
Elk toe	Alasmidonta marginata
Slipper shell	Alasmidonta viridis
White heel-splitter	Lasmigona complanata
Fluted shell	Lasmigona costata
Washboard	Megalonaias nervosa
Pistol-grip	Tritigonia verrucosa

Monkey-face	Quadrula metanevra
Pimple-back	Quadrula pustulosa
Three-ridge	Amblema plicata
Wabash pig-toe	Fusconaia flava
Round pig-toe	Pleurobema sintoxia
Lady-finger	Elliptio dilatata
Three-horned warty-back	Obliquaria reflexa
Mucket	Actinonaias ligamentina carinata
Ellipse	Venustaconcha ellipsiformis ellipsiformis
Butterfly	Ellipsaria lineolata
Deer-toe	Truncilla truncata
Fawn's foot	Truncilla donaciformis
Scale shell	Leptodea leptodon
Fragile paper shell	Leptodea fragilis
Pink heel-splitter	Potamilis alatus
Pink paper shell	Potamilus ohioensis
Liliput shell	Toxolasma parvus
Black sand shell	Ligumia recta
Slough sand shell	Lampsilis teres teres
Yellow sand shell	Lampsilis teres anodontoides
Fat mucket	Lampsilis radiata luteola
Pink mucket	Lampsilis abrupta
Pocketbook	Lampsilis ventricosa
Britt's mussel	Lampsilis reeviana brittsi

Table 3: Crayfish found within the Big River Basin.

Common Name	**Scientific Name**
Freckled crayfish	C. maculatus
Belted crayfish	Orconectes harrisoni

Woodland crayfish	O. hylas
Golden crayfish	O. luteus
Spothanded crayfish	O. punctimanus
Northern crayfish	O. virilis
Saddledback Crayfish	O. medius
Devil crayfish	Cambarus diogenes

Table 4: Species List for Washington County. Federal or state listed Threatened or endangered species are in bold.

Amphibians

* BULLFROG (RANA CATESBEIANA)
* COMMON MUDPUPPY (NECTURUS MACULOSUS MACULOSUS)
* FROG, BLANCHARD'S CRICKET (ACRIS CREPITANS BLANCHARDI)
* FROG, GREEN (RANA CLAMITANS MELANOTA)
* FROG, PICKEREL (RANA PALUSTRIS)
* FROG, SOUTHERN LEOPARD (RANA SPHENOCEPHALA UTRICULARIA)
* NEWT, CENTRAL (NOTOPHTHALMUS VIRIDESCENS LOUISIANENSIS)
* PEEPER, NORTHERN SPRING (PSEUDACRIS CRUCIFER CRUCIFER)
* SALAMANDER, CAVE (EURYCEA LUCIFUGA)
* SALAMANDER, DARK-SIDED (EURYCEA LONGICAUDA MELANOPLEURA)
* SALAMANDER, FOUR-TOED (HEMIDACTYLIUM SCUTATUM)
* SALAMANDER, LONG-TAILED (EURYCEA LONGICAUDA LONGICAUDA)
* SALAMANDER, MARBLED (AMBYSTOMA OPACUM)
* SALAMANDER, SOUTHERN RED-BACKED (PLETHODON SERRATUS)
* SALAMANDER, SPOTTED (AMBYSTOMA MACULATUM)
* SALAMANDER, WESTERN SLIMY (PLETHODON ALBAGULA)
* TOAD, DWARF AMERICAN (BUFO AMERICANUS CHARLESMITHI)
* TOAD, FOWLER'S (BUFO FOWLERI)

Amphipods

* CENTRAL MISSOURI CAVE AMPHIPOD (ALLOCRANGONYX HUBRICHTI)
* ONONDAGA CAVE AMPHIPOD (STYGOBROMUS ONONDAGAENSIS)

Birds

* BLACKBIRD, BREWER'S (EUPHAGUS CYANOCEPHALUS)
* BLACKBIRD, RED-WINGED (AGELAIUS PHOENICUS)
* BLUEBIRD, EASTERN (SIALIA SIALIS SIALIS)
* BOBWHITE, NORTHERN (COLINUS VIRGINIANUS VIRGINIANUS)
* BUNTING, INDIGO (PASSERINA CYANEA)
* BUNTING, SNOW (PLEXTROPHENAX NIVALIS NIVALIS)
* CARDINAL, NORTHERN (CARDINALIS CARDINALIS CARDINALIS)
* CATBIRD, GRAY (DUMETELLA CAROLINENSIS)

* CHAT, YELLOW-BREASTED (ICTERIA VIRENS VIRENS)
* CHICKADEE, CAROLINA (PARUS CAROLINENSIS EXTIMUS)
* CHUCK-WILL'S WIDOW (CAPRIMULGUS CAROLINENSIS)
* COOT, AMERICAN (FULICA AMERICANA AMERICANA)
* COWBIRD, BROWN-HEADED (MOLOTHRUS ATER)
* CROW, AMERICAN (CORVUS BRACHYRHYNCHOS)
* CUCKOO, BLACK-BILLED (COCCYZUS ERYTHROPTHALMUS)
* CUCKOO, YELLOW-BILLED (COCCYZUS AMERICANUS AMERICANUS)
* DICKCISSEL (SPIZA AMERICANA)
* DOVE, MOURNING (ZENAIDA MACROURA CAROLINENSIS)
* DOVE, ROCK (COLUMBA LIVIA)
* DUCK, RING-NECKED (ATHYA COLLARIS)
* DUCK, RUDDY (OXYURA JAMAICENSIS RUBIDA)
* DUCK, WOOD (AIX SPONSA)
* EGRET, CATTLE (BULBULCUS IBIS IBIS)
* FINCH, PURPLE (CARPODACUS PURPUREUS PURPUREUS)
* FLICKER, NORTHERN (COLAPTES AURATUS AURATUS)
* FLYCATCHER, ACADIAN (EMPIDONAX VIRESCENS)
* FLYCATCHER, GREAT CRESTED (MYIARCHUS CRINITUS)
* FLYCATCHER, SCISSOR-TAILED (MUSCIVORA FORFICATA)
* FLYCATCHER, WILLOW (EMPIDONAX TRAILII)
* GADWALL (ANAS STREPERA)
* GNATCATCHER, BLUE-GRAY (POLIOPTILA CAERULEA CAERULEA)
* GOLDENEYE, COMMON (BUCEPHALA CLANGULA AMERICANA)
* GOLDFINCH, AMERICAN (CARDUELIS TRISTIS TRISTIS)
* GRACKLE, COMMON (QUISCALUS QUISCULA)
* GREBE, HORNED (PODICEPS AURITUS)
* GROSBEAK, BLUE (PASSERINA CAERULEA CAERULEA)
* GROSBEAK, EVENING (COCCOTHRAUSTES VESPERTINUS VESPERTINUS)
* GROUSE, RUFFED (BONASA UMBELLUS MEDIANA)
* **HARRIER, NORTHERN (CIRCUS CYANEUS)**
* HAWK, BROAD-WINGED (BUTEO PLATYPTERUS PLATYPTERUS)
* HAWK, COOPER'S (ACCIPITER COOPERII)
* HAWK, RED-SHOULDERED (BUTEO LINEATUS LINEATUS)
* HAWK, RED-TAILED (BUTEO JAMAICENSIS BOREALIS)
* HAWK, ROUGH-LEGGED (BUTEO LAGOPUS SANCTIJOHANNIS)
* HAWK, SHARP-SHINNED (ACCIPITER STRIATUS VELOX)
* HERON, GREAT BLUE (ARDEA HERODIAS)
* HERON, GREEN (BUTORIDES VIRESCENS)
* HUMMINGBIRD, RUBY-THROATED (ARCHILOCHUS COLUBRIS)
* JAY, BLUE (CYANOCITTA CRISTATA CRISTATA)
* JUNCO, DARK-EYED (JUNCO HYEMALIS)
* KESTREL, AMERICAN (FALCO SPARVERIUS SPARVERIUS)
* KILLDEER (CHARADRIUS VOCIFERUS)
* KINGBIRD, EASTERN (TYRANNUS TYRANNUS)
* KINGBIRD, WESTERN (TYRANNUS VERTICALIS)
* KINGFISHER, BELTED (MEGACERYLE ALCYON ALCYON)

* KINGLET, GOLDEN-CROWNED (REGULUS SATRAPA SATRAPA)
* MALLARD (ANAS PLATYRHYNCHOS PLATYRHYNCHOS)
* MARTIN, PURPLE (PROGNE SUBIS SUBIS)
* MEADOWLARK, EASTERN (STURNELLA MAGNA)
* MEADOWLARK, WESTERN (STURNELLA NEGLECTA)
* MERGANSER, HOODED (LOPHODYTES CUCULLATUS)
* MOCKINGBIRD, NORTHERN (MIMUS POLYGLOTTOS POLYGLOTTOS)
* NIGHTHAWK, COMMON (CHORDEILES MINOR)
* NUTHATCH, RED-BREASTED (SITTA CANADENSIS)
* NUTHATCH, WHITE-BREASTED (SITTA CAROLINENSIS CAROLINENSIS)
* ORIOLE, BALTIMORE (ICTERUS GALBULA)
* ORIOLE, ORCHARD (ICTERUS SPURIUS)
* OVENBIRD (SEIURUS AUROCAPILLUS)
* OWL, BARRED (STRIX VARIA)
* OWL, EASTERN SCREECH (OTUS ASIO ASIO)
* OWL, GREAT HORNED (BUBO VIRGINIANUS)
* PARULA, NORTHERN (PARULA AMERICANA)
* PEWEE, EASTERN WOOD- (CONTOPUS VIRENS)
* PHOEBE, EASTERN (SAYORNIS PHOEBE)
* PINTAIL, NORTHERN (ANAS ACUTA)
* REDHEAD (ATHYA AMERICANA)
* REDSTART, AMERICAN (SETOPHAGA RUTICILLA)
* ROBIN, AMERICAN (TURDUS MIGRATORIUS MIGRATORIUS)
* SAPSUCKER, YELLOW-BELLIED (SPHYRAPICUS VARIUS)
* SCAUP, LESSER (ATHYA AFFINIS)
* SCOTER, SURF (MELANITTA PERSPICILLATA)
* SHOVELER, NORTHERN (ANAS CLYPEATA)
* SHRIKE, LOGGERHEAD (LANIUS LUDOVICIANUS MIGRANS)
* SORA (PORZANA CAROLINA)
* SPARROW, AMERICAN TREE (SPIZELLA ARBOREA ARBOREA)
* SPARROW, FIELD (SPIZELLA PUSILLA PUSILLA)
* SPARROW, FOX (PASSERELLA ILIACA ILIACA)
* SPARROW, GRASSHOPPER (AMMODRAMUS SAVANNARUM PRATENSIS)
* SPARROW, HOUSE (PASSER DOMESTICUS)
* SPARROW, SONG (MELOSPIZA MELODIA MELODIA)
* SPARROW, SWAMP (MELOSPIZA GEORGIANA GEORGIANA)
* SPARROW, WHITE-CROWNED (ZONOTRICHA LEUCOPHRYS LEUCOPHRYS)
* SPARROW, WHITE-THROATED (ZONOTRICHA ALBICOLLIS)
* STARLING, EUROPEAN (STURNUS VULGARIS VULGARIS)
* SWALLOW, BARN (HIRUNDO RUSTICO)
* SWALLOW, NORTHERN ROUGH-WINGED (STELGIDOPTERYX SERRIPENNIS)
* SWALLOW, TREE (IRIDOPROCNE BICOLOR)
* SWIFT, CHIMNEY (CHAETURA PELAGICA)
* TANAGER, SCARLET (PIRANGA OLIVACEA)
* TANAGER, SUMMER (PIRANGA RUBRA)
* TEAL, BLUE-WINGED (ANAS DISCORS DISCORS)
* TEAL, GREEN-WINGED (ANAS CRECCA)

* THRASHER, BROWN (TOXOSTOMA RUFUM RUFUM)
* THRUSH, WOOD (HYLOCICHLA MUSTELINA)
* TITMOUSE, TUFTED (PARUS BICOLOR)
* TOWHEE, EASTERN (PIPILO ERYTHROPHTHALMUS)
* TURKEY, WILD (MELEAGRIS GALLOPAVO SILVESTRIS)
* VIREO, PHILADELPHIA (VIREO PHILADELPHICUS)
* VIREO, RED-EYED (VIREO OLIVACEUS)
* VIREO, WARBLING (VIREO GILVUS)
* VIREO, WHITE-EYED (VIREO GRISEUS)
* VIREO, YELLOW-THROATED (VIREO FLAVIFRONS)
* VULTURE, TURKEY (CATHARTES AURA SEPTENTRIONALIS)
* WARBLER, BLACK-AND-WHITE (MNIOTILTA VARIA)
* WARBLER, BLUE-WINGED (VERMIVORA PINUS)
* WARBLER, CERULEAN (DENDROICA CERULEA)
* WARBLER, GOLDEN-WINGED (VERMIVORA CHRYSOPTERA)
* WARBLER, HOODED (WILSONIA CITRINA)
* WARBLER, KENTUCKY (OPORORNIS FORMOSUS)
* WARBLER, PINE (DENDROICA PINUS PINUS)
* WARBLER, PRAIRIE (DENDROICA DISCOLOR)
* WARBLER, PROTHONOTARY (PROTONOTARIA CITREA)
* WARBLER, WORM-EATING (HELMITHEROS VERMIVORUS)
* WARBLER, YELLOW (DENDROICA PETECHIA)
* WARBLER, YELLOW-RUMPED (DENDROICA CORONATA)
* WARBLER, YELLOW-THROATED (DENDROICA DOMINICA)
* WATERTHRUSH, LOUISIANA (SEIURUS MOTACILLA)
* WAXWING, CEDAR (BOMBYCILLA CEDRORUM)
* WHIP-POOR-WILL (CAPRIMULGUS VOCIFERUS)
* WIGEON, AMERICAN (ANAS AMERICANA)
* WOODCOCK, AMERICAN (PHILOHELA MINOR)
* WOODPECKER, DOWNY (PICOIDES PUBESCENS PUBESCENS)
* WOODPECKER, HAIRY (PICOIDES VILLOSUS)
* WOODPECKER, PILEATED (DRYOCOPUS PILEATUS)
* WOODPECKER, RED-BELLIED (MELANERPES CAROLINUS)
* WOODPECKER, RED-HEADED (MELANERPES ERYTHROCEPHALUS)
* WREN, BEWICK'S (TROGLODYTES BEWICKII BEWICKII)
* WREN, CAROLINA (THRYOTHORUS LUDOVICIANUS LUDOVICIANUS)
* WREN, HOUSE (TROGLODYTES AEDON PARKMANII)
* YELLOWTHROAT, COMMON (GEOTHLYPIS TRICHAS BRACHIDACTYLUS)

Crayfish

* CRAYFISH, BELTED (ORCONECTES HARRISONI)
* CRAYFISH, DEVIL (CAMBARUS DIOGENES)
* CRAYFISH, FRECKLED (CAMBARUS MACULATUS)
* CRAYFISH, GOLDEN (ORCONECTES LUTEUS)
* CRAYFISH, SADDLEBACKED (ORCONECTES MEDIUS)
* CRAYFISH, SPOTHANDED (ORCONECTES PUNCTIMANUS)
* CRAYFISH, VIRILE (ORCONECTES VIRILIS)
* CRAYFISH, WOODLAND (ORCONECTES HYLAS)

Fish

* BASS, LARGEMOUTH (MICROPTERUS SALMOIDES)
* BASS, ROCK (AMBLOPLITES RUPESTRIS)
* BASS, SMALLMOUTH (MICROPTERUS DOLOMIEU)
* BLUEGILL (LEPOMIS MACROCHIRUS)
* BULLHEAD, YELLOW (AMEIURUS NATALIS)
* CHUB, BIGEYE (NOTROPIS AMBLOPS)
* CHUB, CREEK (SEMOTILUS ATROMACULATUS)
* CHUB, GRAVEL (ERIMYSTAX X-PUNCTATA)
* CHUB, HORNYHEAD (NOCOMIS BIGUTTATUS)
* CHUBSUCKER, CREEK (ERIMYZON OBLONGUS)
* DACE, SOUTHERN REDBELLY (PHOXINUS ERYTHROGASTER)
* DARTER, BANDED (ETHEOSTOMA ZONALE)
* DARTER, FANTAIL (ETHEOSTOMA FLABELLARE)
* DARTER, GREENSIDE (ETHEOSTOMA BLENNIOIDES)
* DARTER, JOHNNY (ETHEOSTOMA NIGRUM)
* DARTER, MISSOURI SADDLED (ETHEOSTOMA TETRAZONUM)
* DARTER, ORANGETHROAT (ETHEOSTOMA SPECTABILE)
* DARTER, RAINBOW (ETHEOSTOMA CAERULEUM)
* LAMPREY, LEAST BROOK (LAMPETRA AEPYPTERA)
* LOGPERCH (PERCINA CAPRODES)
* MADTOM, SLENDER (NOTURUS EXILIS)
* MINNOW, BLUNTNOSE (PIMEPHALES NOTATUS)
* MINNOW, FATHEAD (PIMEPHALES PROMELAS)
* MINNOW, OZARK (NOTROPIS NUBILA)
* MINNOW, SILVERJAW (NOTROPIS BUCCATUS)
* PICKEREL, GRASS (ESOX AMERICANUS)
* REDHORSE, BLACK (MOXOSTOMA DUQUESNEI)
* REDHORSE, GOLDEN (MOXOSTOMA ERYTHRURUM)
* SCULPIN, BANDED (COTTUS CAROLINAE)
* SCULPIN, MOTTLED (COTTUS BAIRDI)
* SHINER, BIGEYE (NOTROPIS BOOPS)
* SHINER, BLEEDING (LUXILUS ZONATUS)
* SHINER, CARMINE (NOTROPIS PERCOBROMUS)
* SHINER, GOLDEN (NOTEMIGONUS CRYSOLEUCAS)
* SHINER, MIMIC (NOTROPIS VOLUCELLUS)
* SHINER, REDFIN (LYTHRURUS UMBRATILIS)
* SHINER, SAND (NOTROPIS STRAMINEUS)
* SHINER, SPOTFIN (CYPRINELLA SPILOPTERA)
* SHINER, STEELCOLOR (CYPRINELLA WHIPPLEI)
* SHINER, STRIPED (LUXILUS CHRYSOCEPHALUS)
* SHINER, WEDGESPOT (NOTROPIS GREENEI)
* SILVERSIDE, BROOK (LABIDESTHES SICCULUS)
* STONECAT (NOTURUS FLAVUS)
* STONEROLLER, CENTRAL (CAMPOSTOMA PULLUM)
* STONEROLLER, LARGESCALE (CAMPOSTOMA OLIGOLEPIS)
* STUDFISH, NORTHERN (FUNDULUS CATENATUS)

* SUCKER, NORTHERN HOG (HYPENTELIUM NIGRICANS)
* SUNFISH, GREEN (LEPOMIS CYANELLUS)
* SUNFISH, LONGEAR (LEPOMIS MEGALOTIS)
* SUNFISH, REDEAR (LEPOMIS MICROLOPHUS)
* TOPMINNOW, BLACKSPOTTED (FUNDULUS OLIVACEOUS)
* A HEPTAGENIID MAYFLY (STENONEMA BEDNARIKI)
* WESTFALL'S SNAKETAIL (OPHIOGOMPHUS WESTFALLI)

Mammals

* ARMADILLO, NINE-BANDED (DASYPUS NOVEMCINCTUS MEXICANUS)
* BAT, BIG BROWN (EPTESICUS FUSCUS FUSCUS)
* BAT, EVENING (NYCTICEIUS HUMERALIS HUMERALIS)
* **BAT, GRAY (MYOTIS GRISESCENS)**
* BAT, HOARY (LASIURUS CINEREA CINEREA)
* **BAT, INDIANA (MYOTIS SODALIS)**
* BAT, LITTLE BROWN (MYOTIS LUCIFUGUS LUCIFUGUS)
* BAT, NORTHERN (MYOTIS SEPTENTRIONALIS)
* BAT, RED (LASIURUS BOREALIS BOREALIS)
* BAT, SILVER-HAIRED (LASIONYCTERIS NOCTIVAGANS)
* BEAR, BLACK (URSUS AMERICANUS AMERICANUS)
* BEAVER (CASTOR CANADENSIS CAROLINENSIS)
* BOBCAT (LYNX RUFUS RUFUS)
* CHIPMUNK, EASTERN (TAMIAS STRIATUS GRISEUS)
* COYOTE (CANIS LATRANS FRUSTROR)
* DEER, WHITE-TAILED (ODOCOILEUS VIRGINIANUS)
* FOX, GRAY (UROCYON CINEREOARGENTEUS)
* FOX, RED (VULPES VULPES FULVA)
* MINK (MUSTELA VISON LETIFERA)
* MOUSE, HOUSE (MUS MUSCULUS DOMESTICUS)
* MUSKRAT (ONDATRA ZIBETHICUS)
* MYOTIS, EASTERN SMALL-FOOTED (MYOTIS LEIBII)
* OPOSSUM, VIRGINIA (DIDELPHIS VIRGINIANA VIRGINIANA)
* OTTER, RIVER (LONTRA CANADENSIS)
* PIPISTRELLE, EASTERN (PIPISTRELLUS SUBFLAVUS)
* RABBIT, EASTERN COTTONTAIL (SYLVILAGUS FLORIDANUS ALACER)
* RACCOON (PROCYON LOTOR HIRTUS)
* RAT, EASTERN WOOD (NEOTOMA FLORIDANA)
* RAT, NORWAY (RATTUS NORVEGICUS NORVEGICUS)
* **SKUNK, PLAINS SPOTTED (SPILOGALE PUTORIUS)**
* SKUNK, STRIPED (MEPHITIS MEPHITIS AVIA)
* SQUIRREL, FOX (SCIURUS NIGER RUFIVENTER)
* SQUIRREL, GRAY (SCIURUS CAROLINENSIS CAROLINENSIS)
* SQUIRREL, SOUTHERN FLYING (GLAUCOMYS VOLANS)
* WOODCHUCK (MARMOTA MONAX MONAX)

Flora

* BIG-BEAKED WOODSY MOSS (PLAGIOMNIUM ROSTRATUM)
* BUTTERNUT (JUGLANS CINEREA)
* COMMON LADIES' TRESSES (SPIRANTHES CERNUA)

* CRESTED CORAL ROOT (HEXALECTRIS SPICATA SPICATA)
* GATTINGER'S GOLDENROD (SOLIDAGO GATTINGERI)
* GRASS-OF-PARNASSUS (PARNASSIA GRANDIFOLIA)
* HEART-LEAVED PLANTAIN (PLANTAGO CORDATA)
* LARGE TWAYBLADE (LIPARIS LILIIFOLIA)
* LATE CORAL ROOT (CORALLORHIZA ODONTORHIZA)
* LITTLE LADIES' TRESSES (SPIRANTHES TUBEROSA)
* OVAL LADIES' TRESSES (SPIRANTHES OVALIS EROSTELLATA)
* ROCK CRESS (ARABIS MISSOURIENSIS)
* ROYAL CATCHFLY (SILENE REGIA)
* SAND GRAPE (VITIS RUPESTRIS)
* SHINING LADIES' TRESSES (SPIRANTHES LUCIDA)
* SHOWY ORCHIS (GALEARIS SPECTABILIS)
* SLENDER LADIES' TRESSES (SPIRANTHES LACERA)
* SULLIVANT (ORANGE) CONEFLOWER (RUDBECKIA FULGIDA SULLIVANTII)
* WEAKSTALK BULRUSH (SCHOENOPLECTUS PURSHIANUS)
* WILD SWEET WILLIAM (PHLOX MACULATA PYRAMIDALIS)
* YELLOW LADY'S SLIPPER (CYPRIPEDIUM CALCEOLUS)

Reptiles

* COACHWHIP, EASTERN (MASTICOPHIS FLAGELLUM FLAGELLUM)
* COPPERHEAD, OSAGE (AGKISTRODON CONTORTRIX PHAEOGASTER)
* COTTONMOUTH, WESTERN (AGKISTRODON PISCIVORUS LEUCOSTOMA)
* KINGSNAKE, PRAIRIE (LAMPROPELTIS CALLIGASTER CALLIGASTER)
* KINGSNAKE, SPECKLED (LAMPROPELTIS GETULUS HOLBROOKI)
* LIZARD, EASTERN COLLARED (CROTAPHYTUS COLLARIS)
* LIZARD, NORTHERN FENCE (SCELOPORUS UNDULATUS HYACINTHINUS)
* LIZARD, WESTERN SLENDER GLASS (OPHISAURUS ATTENUATUS ATTENUATUS)
* RACER, EASTERN YELLOW-BELLIED (COLUBER CONSTRICTOR FLAVIVENTRIS)
* RACERUNNER, EASTERN SIX-LINED (CNEMIDOPHORUS SEXLINEATUS SEXLINEATUS)
* RACERUNNER, PRAIRIE (CNEMIDOPHORUS SEXLINEATUS VIRIDIS)
* RATTLESNAKE, TIMBER (CROTALUS HORRIDUS)
* RATTLESNAKE, WESTERN PIGMY (SISTRURUS MILIARIUS STRECKERI)
* SKINK, BROAD-HEADED (EUMECES LATICEPS)
* SKINK, COMMON FIVE-LINED (EUMECES FASCIATUS)
* SKINK, GROUND (SCINCELLA LATERALIS)
* SLIDER, RED-EARED (TRACHEMYS SCRIPTA ELEGANS)
* SNAKE, BLACK RAT (ELAPHE OBSOLETA OBSOLETA)
* SNAKE, EASTERN GARTER (THAMNOPHIS SIRTALIS SIRTALIS)
* SNAKE, EASTERN HOG-NOSED (HETERODON PLATIRHINOS)
* SNAKE, FLAT-HEADED (TANTILLA GRACILIS)
* SNAKE, GREAT PLAINS RAT (ELAPHE GUTTATA EMORYI)
* SNAKE, MIDLAND BROWN (STORERIA DEKAYI WRIGHTORUM)
* SNAKE, NORTHERN RED-BELLIED (STORERIA OCCIPITOMACULATA OCCIPITOMACULATA)
* SNAKE, NORTHERN WATER (NERODIA SIPEDON SIPEDON)
* SNAKE, PRAIRIE RING-NECKED (DIADOPHIS PUNCTATUS ARNYI)
* SNAKE, RED MILK (LAMPROPELTIS TRIANGULUM SYSPILA)
* SNAKE, ROUGH GREEN (OPHEODRYS AESTIVUS AESTIVUS)

* SNAKE, WESTERN EARTH (VIRGINIA VALERIAE ELEGANS)
* SNAKE, WESTERN RIBBON (THAMNOPHIS PROXIMUS PROXIMUS)
* SNAKE, WESTERN WORM (CARPHOPHIS VERMIS)
* SOFTSHELL, WESTERN SPINY (APALONE SPINIFERA HARTWEGI)
* TURTLE, COMMON MAP (GRAPTEMYS GEOGRAPHICA)
* TURTLE, COMMON MUSK (STERNOTHERUS ODORATUS)
* TURTLE, COMMON SNAPPING (CHELYDRA SERPENTINA SERPENTINA)
* TURTLE, ORNATE BOX (TERRAPENE ORNATA ORNATA)
* TURTLE, THREE-TOED BOX (TERRAPENE CAROLINA TRIUNGUIS)
* TURTLE, WESTERN PAINTED (CHRYSEMYS PICTA BELLII)

Table 5. Proposed Sample Designations, Descriptions, and Rationale for the Potosi Site.

Sample ID	Location Description	Depth Intervals	Analyses					Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture	Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-09-SD01-S	Mill Creek	0-6	x		x	x							
PO-09-SD02-S	Mill Creek	0-6	x		x	x							
PO-09-SD03	Tiff Road Tailings Pond	0-6	x		x	x							
PO-09-SD04	Polite Road Tailings Pond	0-6	x		x	x							
PO-09-SD05-S	Mill Creek	0-6	x		x	x							
PO-08-SD06	Shibboleth Tailings Pond	0-6	x		x	x							
PO-08-SD07	Powder Spring Lake	0-6	x		x	x							
PO-08-SD08	Powder Spring Lake	0-6	x		x	x							
PO-08-SD09	Cadet Tailings Pond	0-6	x		x	x							
PO-08-SD10	Hwy 47 Tailings Pond	0-6	x		x	x							

Sample ID	Location Description	Depth Intervals	Analyses					Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture	Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-07-SD11-S	Mill Creek	0-6	x		x	x							
PO-07-SD12-S	Mill Creek	0-6	x		x	x							
PO-07-SD13-S	Mill Creek	0-6	x		x	x							
PO-07-SD14	Midwest Road Tailings Pond	0-6	x		x	x							
PO-06-SD15	Mineral Point Tailings Pond	0-6	x		x	x							
PO-06-SD16	Pond Creek Tailings Pond	0-6	x		x	x							
PO-06-SD17	Pond Creek Tailings Pond	0-6	x		x	x							
PO-06-SD18-S	Pond Creek	0-6	x		x	x							
PO-06-SD19-S	Pond Creek	0-6	x		x	x							
PO-05-SD20	New Diggins Tailing Pond	0-6	x		x	x							
PO-05-SD20-FD	New Diggins Tailing Pond	0-6	x		x	x							

Sample ID	Location Description	Depth Intervals	Analyses						Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture		Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-04-SD21	Mineral Point Tailings Pond	0-6	x		x	x								
PO-04-SD22	Mineral Point Tailings Pond	0-6	x		x	x								
PO-04-SD23	Mineral Point Tailings Pond	0-6	x		x	x								
PO-03-SD24	Settle Mine Tailings Pond	0-6	x		x	x								
PO-03-SD25	Bell Street Tailings Pond	0-6	x		x	x								
PO-03-SD26	Hwy 21 Tailings Pond	0-6	x		x	x								
PO-03-SD27	Hwy East Tailings Pond	0-6	x		x	x								
PO-02-SD28	Hornsey Lake	0-6	x		x	x								
PO-02-SD29	South Mine Tailings pond	0-6	x		x	x								
PO-02-SD30-S	Bates Creek	0-6	x		x	x								
PO-02-SD31-S	Mine A Breton	0-6	x		x	x								

Sample ID	Location Description	Depth Intervals	Analyses						Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture		Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-02-SD32-S	Mine A Breton	0-6	x		x	x								
PO-01-SD33	Gun Club Road Tailings Pond	0-6	x		x	x								
PO-01-SD34-S	Mine A Breton	0-6	x		x	x								
PO-02-SD35-S	Bates Creek-bkg	0-6	x		x	x								
PO-09-SW01-S	Mill Creek	0-12	x				X			x	x	x	x	x
PO-09-SW02-S	Mill Creek	0-12	x				X			x	x	x	x	x
PO-09-SW03	Tiff Road Tailings Pond	0-12	x				X			x	x	x	x	x
PO-09-SW04	Polite Road Tailings Pond	0-12	x				X			x	x	x	x	x
PO-09-SW05-S	Mill Creek	0-12	x				X			x	x	x	x	x
PO-08-SW06	Shibboleth Tailings Pond	0-12	x				X			x	x	x	x	x

Sample ID	Location Description	Depth Intervals	Analyses					Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture	Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-08-SW07	Powder Spring Lake	0-12	x			X			x	x	x	x	x
PO-08-SW08	Powder Spring Lake	0-12	x			X			x	x	x	x	x
PO-08-SW09	Cadet Tailings Pond	0-12	x			X			x	x	x	x	x
PO-08-SW10	Hwy 47 Tailings Pond	0-12	x			X			x	x	x	x	x
PO-07-SW11-S	Mill Creek	0-12	x			X			x	x	x	x	x
PO-07-SW12-S	Mill Creek	0-12	x			X			x	x	x	x	x
PO-07-SW13-S	Mill Creek	0-12	x			X			x	x	x	x	x
PO-07-SW14	Midwest Road Tailings Pond	0-12	x			X			x	x	x	x	x
PO-06-SW15	Mineral Point Tailings Pond	0-12	x			X			x	x	x	x	x
PO-06-SW16	Pond Creek Tailings Pond	0-12	x			X			x	x	x	x	x
PO-06-SW17	Pond Creek Tailings Pond	0-12	x			X			x	x	x	x	x

Sample ID	Location Description	Depth Intervals	Analyses						Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture		Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-06-SW18-S	Pond Creek	0-12	x				X			x	x	x	x	x
PO-06-SW19-S	Pond Creek	0-12	x				X			x	x	x	x	x
PO-05-SW20	New Diggins Tailing Pond	0-12	x				X			x	x	x	x	x
PO-05-SW20-FD	New Diggins Tailing Pond	0-12	x				X			x	x	x	x	x
PO-04-SW21	Mineral Point Tailings Pond	0-12	x				X			x	x	x	x	x
PO-04-SW22	Mineral Point Tailings Pond	0-12	x				X			x	x	x	x	x
PO-04-SW23	Mineral Point Tailings Pond	0-12	x				X			x	x	x	x	x
PO-03-SW24	Settle Mine Tailings Pond	0-12	x				X			x	x	x	x	x
PO-03-SW25	Bell Street Tailings Pond	0-12	x				X			x	x	x	x	x
PO-03-SW26	Hwy 21 Tailings Pond	0-12	x				X			x	x	x	x	x

Sample ID	Location Description	Depth Intervals	Analyses						Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture		Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-03-SW27	Hwy East Tailings Pond	0-12	x							x	x	x	x	x
PO-02-SW28	Hornsey Lake	0-12	x				X			x	x	x	x	x
PO-02-SW29	South Mine Tailings pond	0-12	x				X			x	x	x	x	x
PO-02-SW30-S	Bates Creek	0-12	x				X			x	x	x	x	x
PO-02-SW31-S	Mine A Breton	0-12	x				X			x	x	x	x	x
PO-02-SW32-S	Mine A Breton	0-12	x				X			x	x	x	x	x
PO-01-SW33	Gun Club Road Tailings Pond	0-12	x				X			x	x	x	x	x
PO-01-SW34-S	Mine A Breton	0-12	x				X			x	x	x	x	x
PO-02-SW35-S	Bates Creek - bkg	0-12	x				X			x	x	x	x	x
PO-09-SS01	Tiff Road	0-12	x		X	X			X					
PO-09-SS02	Polite Road	0-12	x		X	X			X					
PO-08-SS03	Hwy 47	0-12	x		X	X			X					

Sample ID	Location Description	Depth Intervals	Analyses					Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture	Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-08-SS04	Hwy 47	0-12	x		X	X		X					
PO-08-SS05	Hwy 47	0-12	x		X	X		X					
PO-08-SS06	Cadet	0-12	x		X	X		X					
PO-07-SS07	Midwest Road	0-12	x		X	X		X					
PO-06-SS08	Trokey Creek	0-12	x		X	X		X					
PO-06-SS09	Pond Creek	0-12	x		X	X		X					
PO-05-SS10	Skiles Road	0-12	x		X	X		X					
PO-05-SS11	Hwy U	0-12	x		X	X		X					
PO-04-SS12	Death Row	0-12	x		X	X		X					
PO-04-SS12-FD	Death Row	0-12	x		X	X		X					
PO-03-SS13	Hwy 21	0-12	x		X	X		X					
PO-03-SS14	Radio Station Rd	0-12	x		X	X		X					
PO-02-SS15	Lakeview	0-12	x		X	X		X					
PO-02-SS16	South Mine	0-12	x		X	X		X					
PO-01-SS17	CR344	0-12	x		X	X		X					
PO-02-SS18	Hwy 21-bkg	0-12	x		X	X		X					
PO-02-SS19	Hwy 8-bkg	0-12	x		X	X		X					
PO-SF-01	TBD		x	X									
PO-SF-02	TBD		x	X									

Sample ID	Location Description	Depth Intervals	Analyses					Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture	Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-SF-03	TBD		x	X									
PO-SF-04	TBD		x	X									
PO-SF-05	TBD		x	X									
PO-SF-06	TBD		x	X									
PO-SF-07	TBD		x	X									
PO-SF-08	TBD		x	X									
PO-SF-09	TBD		x	X									
PO-SF-10	TBD		x	X									
PO-EW-01	TBD	0-12	x										
PO-EW-02	TBD	0-12	x										
PO-EW-03	TBD	0-12	x										
PO-EW-04	TBD	0-12	x										
PO-EW-05	TBD	0-12	x										
PO-EW-06	TBD	0-12	x										
PO-EW-07	TBD	0-12	x										
PO-EW-08	TBD	0-12	x										
PO-EW-09	TBD	0-12	x										
PO-EW-10	TBD	0-12	x										
PO-VG-01	TBD		x										
PO-VG-02	TBD		x										
PO-VG-03	TBD		x										

Sample ID	Location Description	Depth Intervals	Analyses						Field Measurements					
			TAL Metals (in water = total, dissolved, cyanide)	Lipids	TOC	% Moisture		Hardness	pH in soil	Temp.	Cond.	Turb	DO	TDS
PO-VG-04	TBD		x											
PO-VG-05	TBD		x											
PO-VG-06	TBD		x											
PO-VG-07	TBD		x											
PO-VG-08	TBD		x											
PO-VG-09	TBD		x											
PO-VG-10	TBD		x											
PO-CF-01	TBD		x	X										
PO-CF-02	TBD		x	X										
PO-CF-03	TBD		x	X										
PO-CF-04	TBD		x	X										
PO-CF-05	TBD		x	X										

Table 6: SOPs used for each sampling activity.

Sample Collection Activity	Horiba U-10 SOP	Surface Soil SOP	Sediment SOP	Surface Water SOP	Biota SOP	Vegetation SOP	Trimble GPS SOP
Surface Soil		X					X
Sediment			X				X
Sieved Sediment			X				X
Surface Water	X			X			X
Earthworms					X		X
Crayfish					X		X
Fish					X		X
Vegetation						X	X

Table 7. Sample collection containers, requirements, preservation and holding times.

Analysis	Method	Number Containers	Container Type	Preservative	Holding Times
Surface Water/ Pore Water					
TAL Metals ICP-AES+Hg+CN (dissolved)	ILM05.3	1	1 L Polyethylene bottle	Preserve with HNO_3 and cool to 4° C or less	6 months
TAL Metals ICP-AES+Hg+CN (total)	ILM05.3	1	1 L Polyethylene bottle	Preserve with HNO_3 and cool to 4° C or less	6 months
Hardness	Hand	1	Include in TAL bottles	Preserve with HNO_3 and cool to 4° C or less	6 months
Total Organic Carbon	415.1	1	100 ml polyethylene bottle	Preserve with HNO_3 and cool to 4° C or less	28 days
Soil/Sediment/Sieved Sediment					
TAL Metals – ICP-AES TN TAL +Hg+CN	ILM05.3	1	8 oz wide-mouthed glass jar	Cool to 4° C or less	6 months
Total Organic Carbon	SSA Chapter 34	1	4 oz wide-mouthed glass jar	Cool to 4° C or less	6 months
Tissue – Vegetation, Earthworms, Crayfish, Fish					
TAL Metals – ICP-AES TN TAL +Hg+CN	ILM05.3	1	Enclose in aluminum foil and double gallon size Ziploc (need 50g)	Dry ice	6 months

APPENDIX B
FIGURES

Washington County Lead District
Potosi Site - Field Sampling Plan
April 2008

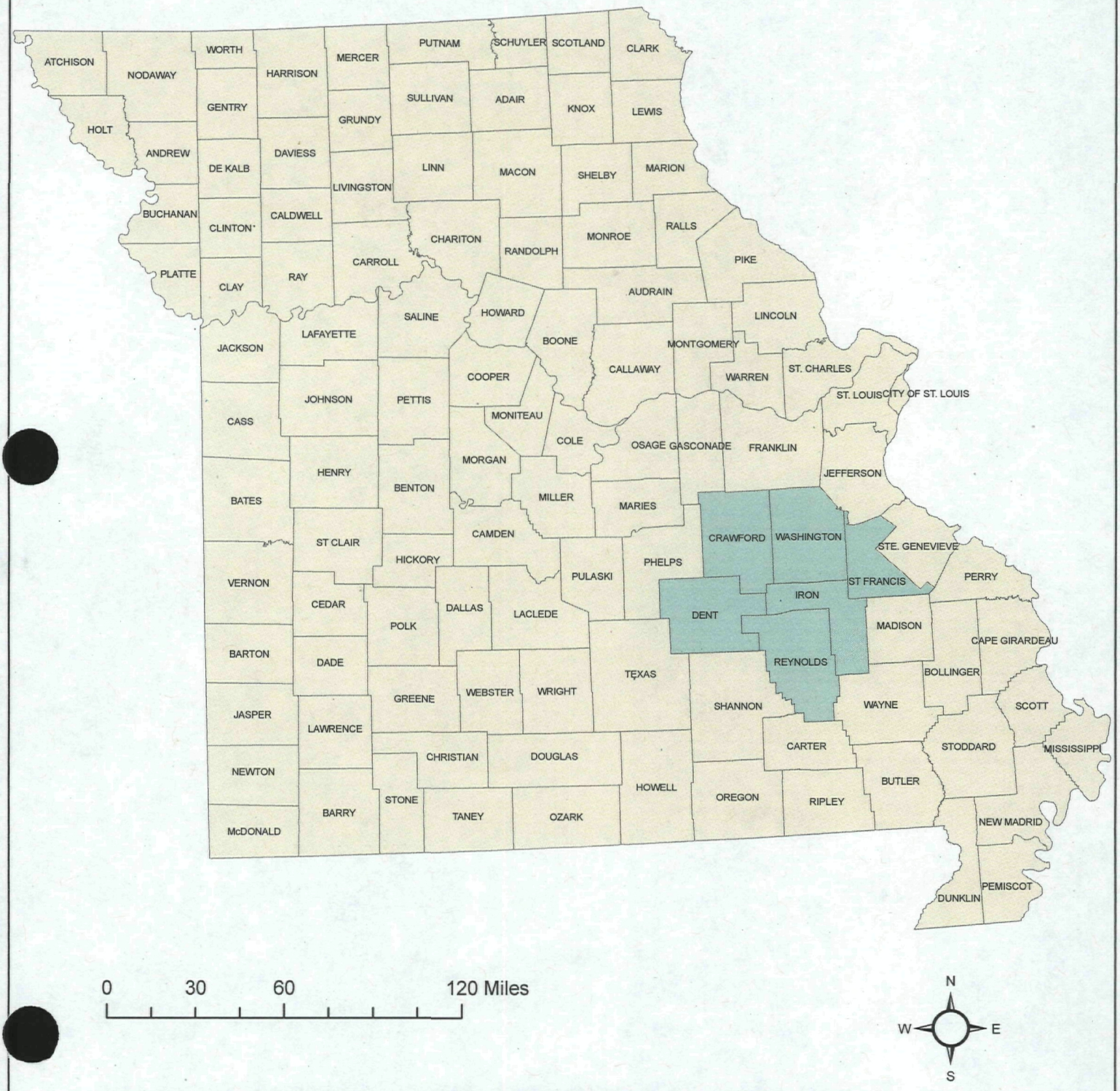
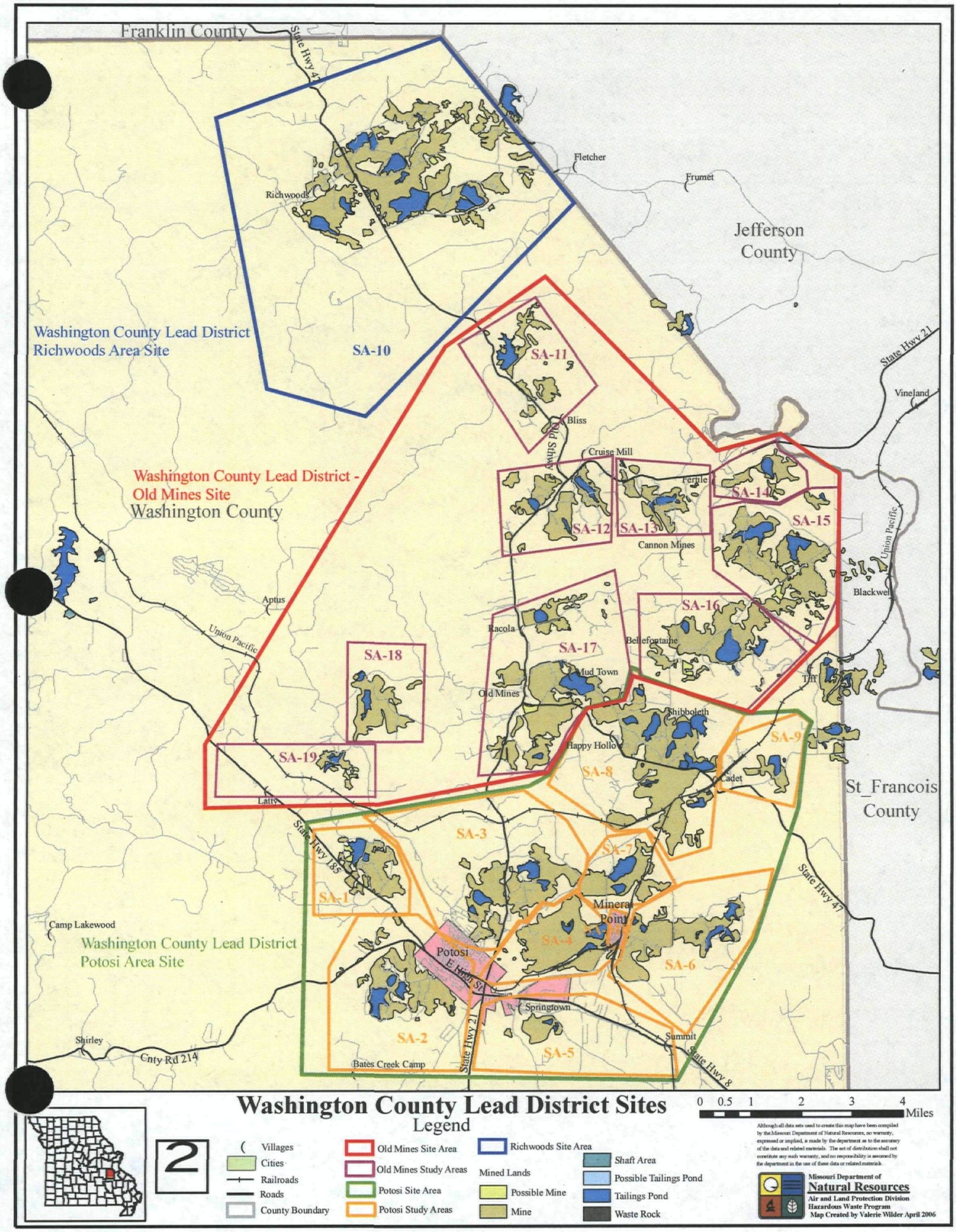


Figure 1. Map of Missouri's Old Lead Belt showing the location of Washington County.


Franklin County
State Hwy 47
Fletcher
Frumet
Richwoods
Jefferson County
Washington County Lead District
Richwoods Area Site
SA-10
SA-11
Bliss
Old Shawmut
Cruise Mill
Washington County Lead District -
Old Mines Site
Washington County
SA-12
SA-13
Fertile
SA-14
SA-15
Cannon Mines
State Hwy 21
Vineland
Union Pacific
Blackwell
Aptus
Union Pacific
SA-16
Racola
Bellefontaine
SA-17
SA-18
Mud Town
Old Mines
Tiff
Shibboleth
SA-19
Happy Hollow
SA-9
SA-8
Cadet
St_Francois County
Latty
State Hwy 185
SA-3
SA-7
SA-1
State Hwy 47
Camp Lakewood
Mineral Point
Washington County Lead District
Potosi Area Site
Potosi
E High St
SA-4
SA-6
Springtown
Shirley
Cnty Rd 214
SA-2
State Hwy 21
SA-5
Summit
State Hwy 8
Bates Creek Camp
Washington County Lead District Sites
Legend
2
Villages
Cities
Railroads
Roads
County Boundary
Old Mines Site Area
Old Mines Study Areas
Potosi Site Area
Potosi Study Areas
Richwoods Site Area
Mined Lands
Possible Mine
Mine
Shaft Area
Possible Tailings Pond
Tailings Pond
Waste Rock
0 0.5 1 2 3 4 Miles
Although all data sets used to create this map have been compiled by the Missouri Department of Natural Resources, no warranty, expressed or implied, is made by the department as to the accuracy of the data and related materials. The act of distribution shall not constitute any such warranty, and no responsibility is assumed by the department in the use of these data or related materials.
Missouri Department of
Natural Resources
Air and Land Protection Division
Hazardous Waste Program
Map Created by Valerie Wilder April 2006

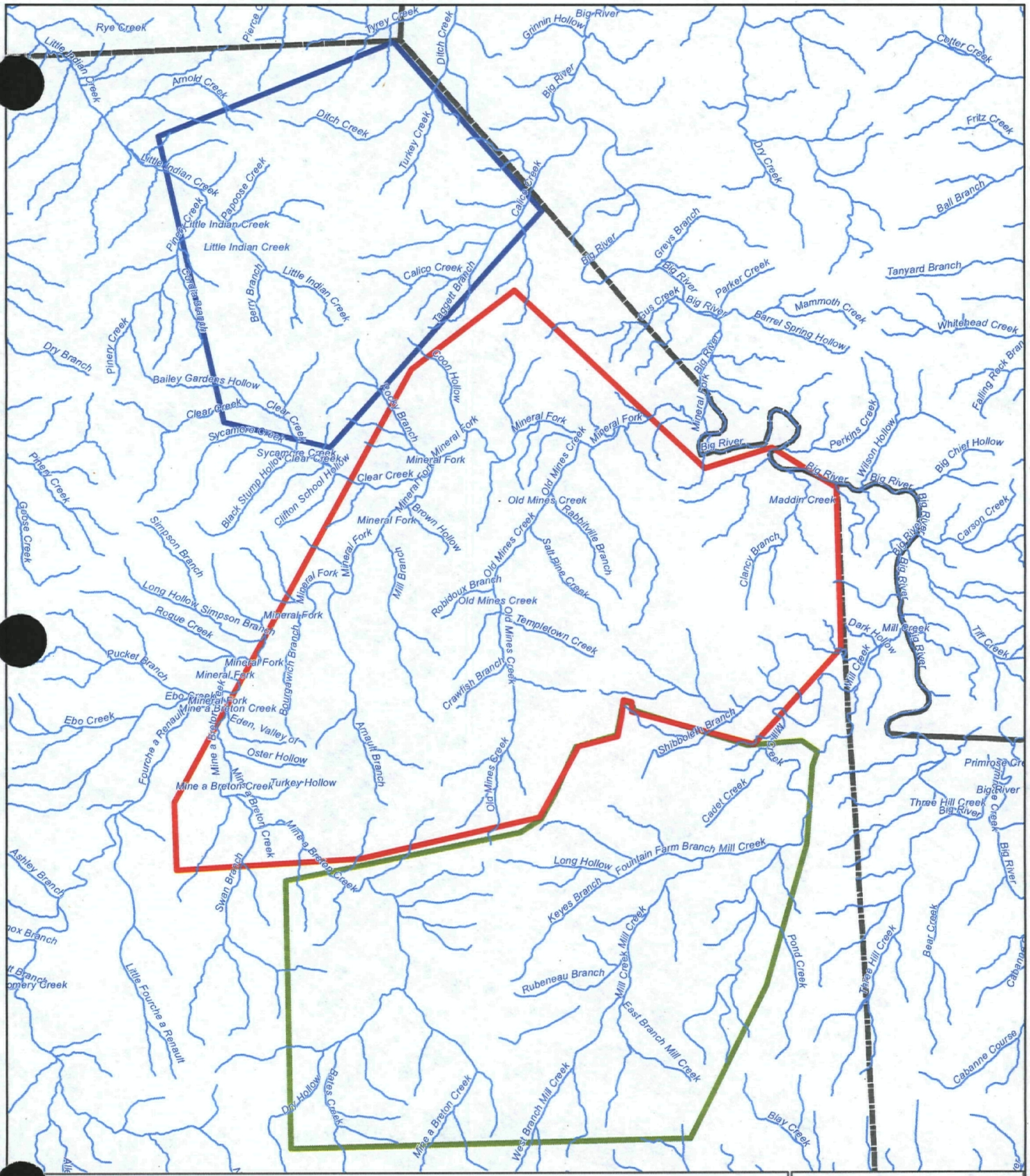


Figure 3: Streams and tributaries within the WCLD site

EPA Region 7

0 1.5 3 Miles

Source of Lead District data: MoDNR

streams/tribs
Richwoods Site Area
Old Mines Site Area
Potosi Site Area
county boundary

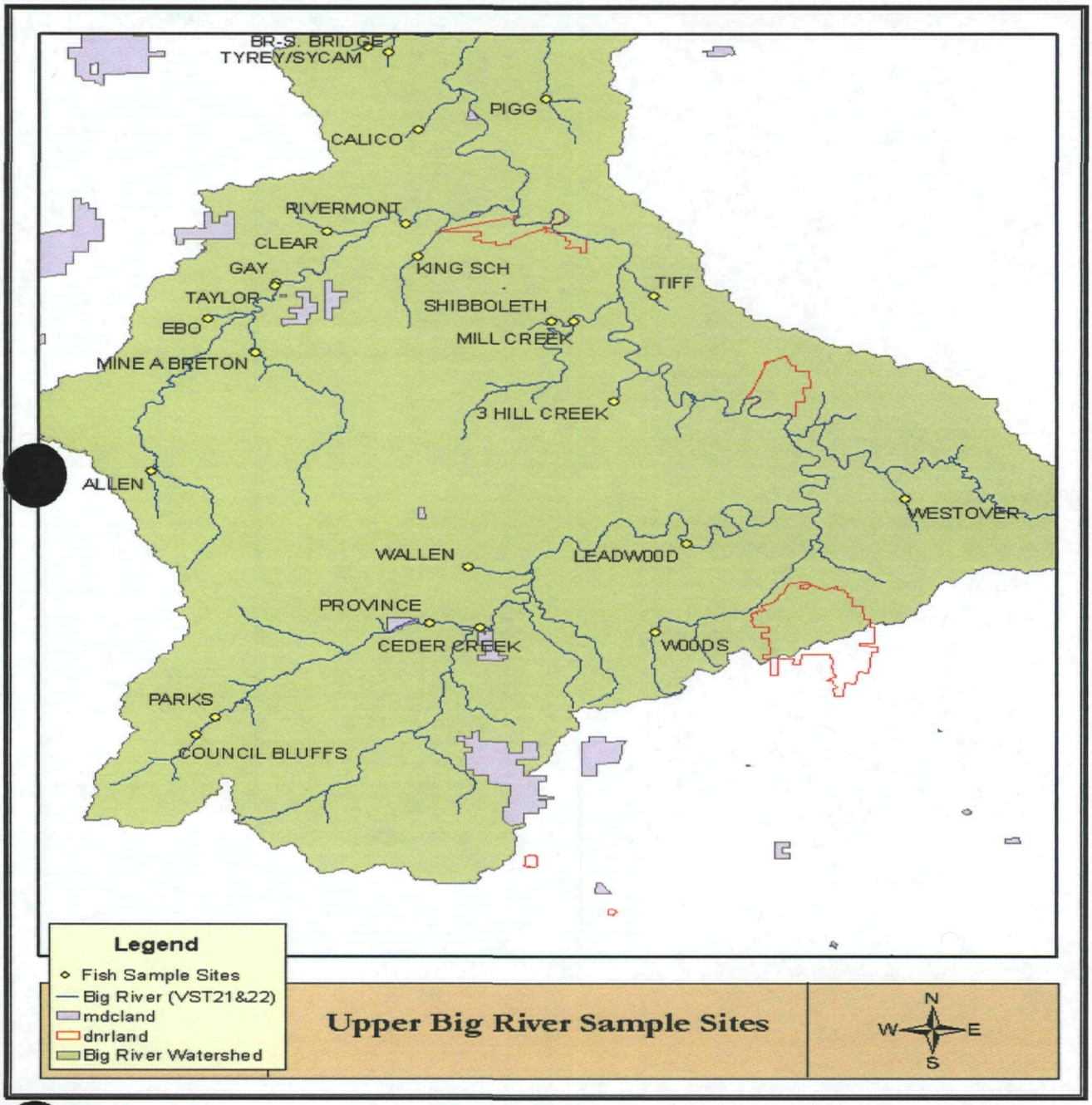


Figure 4. Upper Big River Fish Sampling Locations.

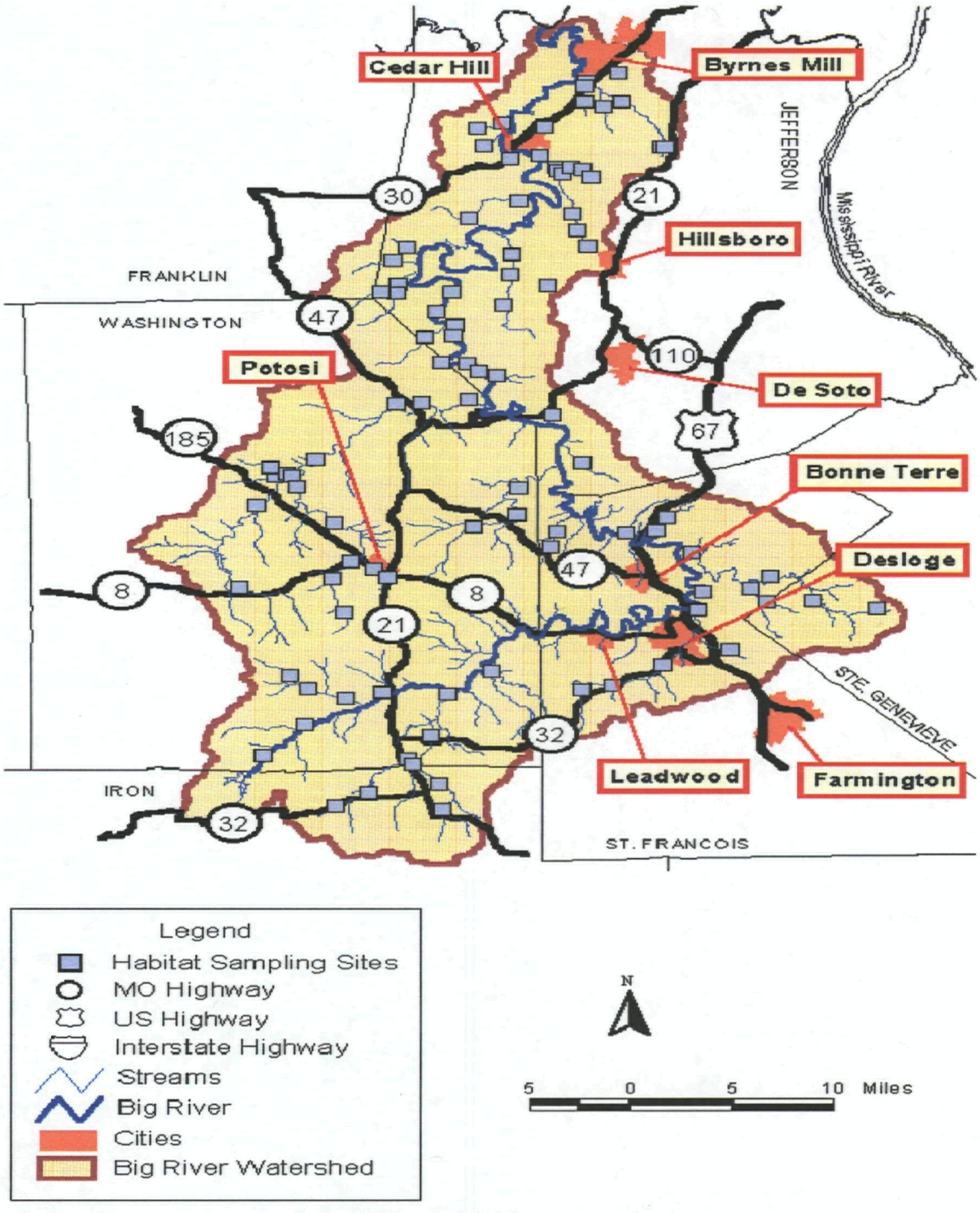
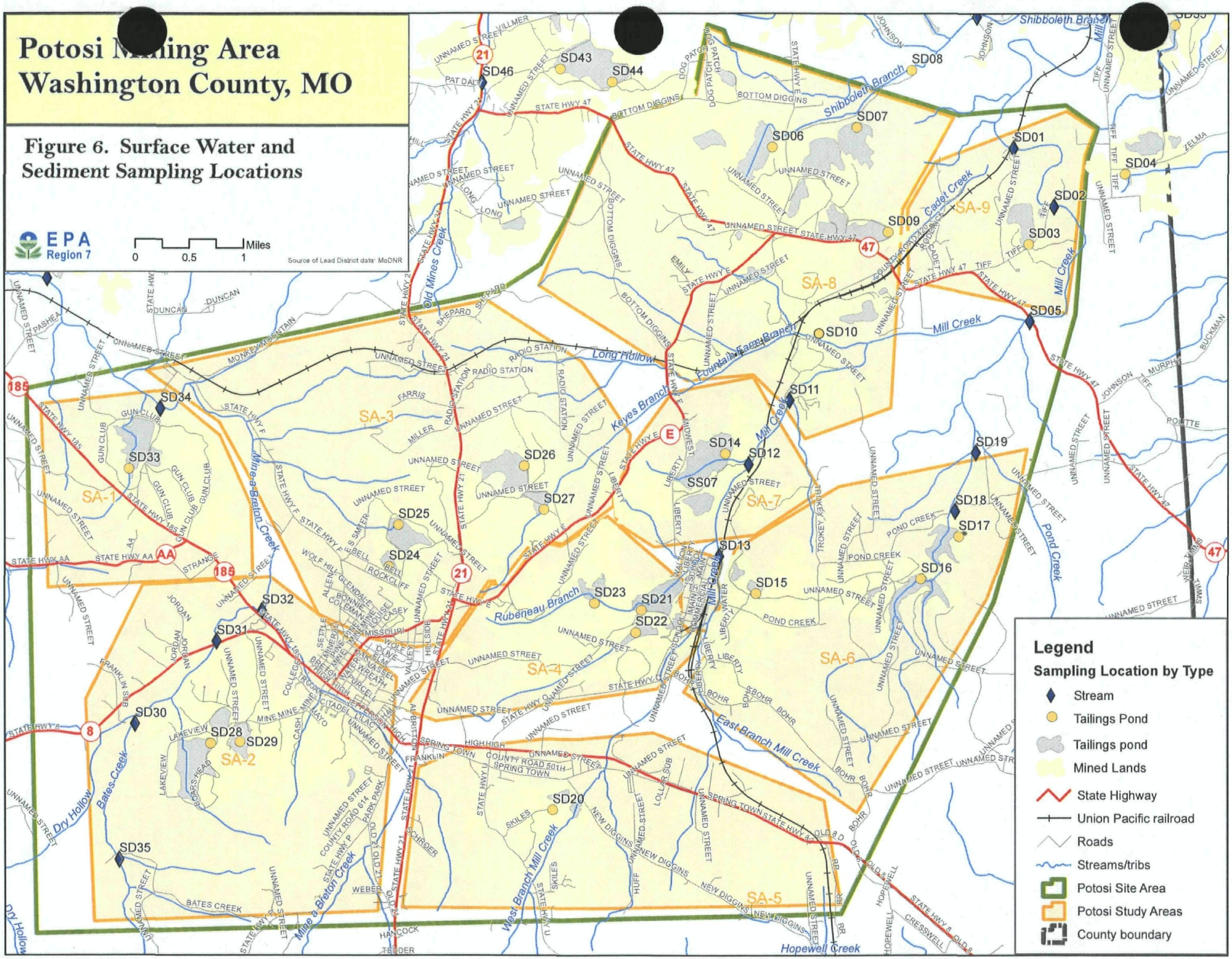


Figure 5: Habitat Sampling Sites in the Big River Basin.

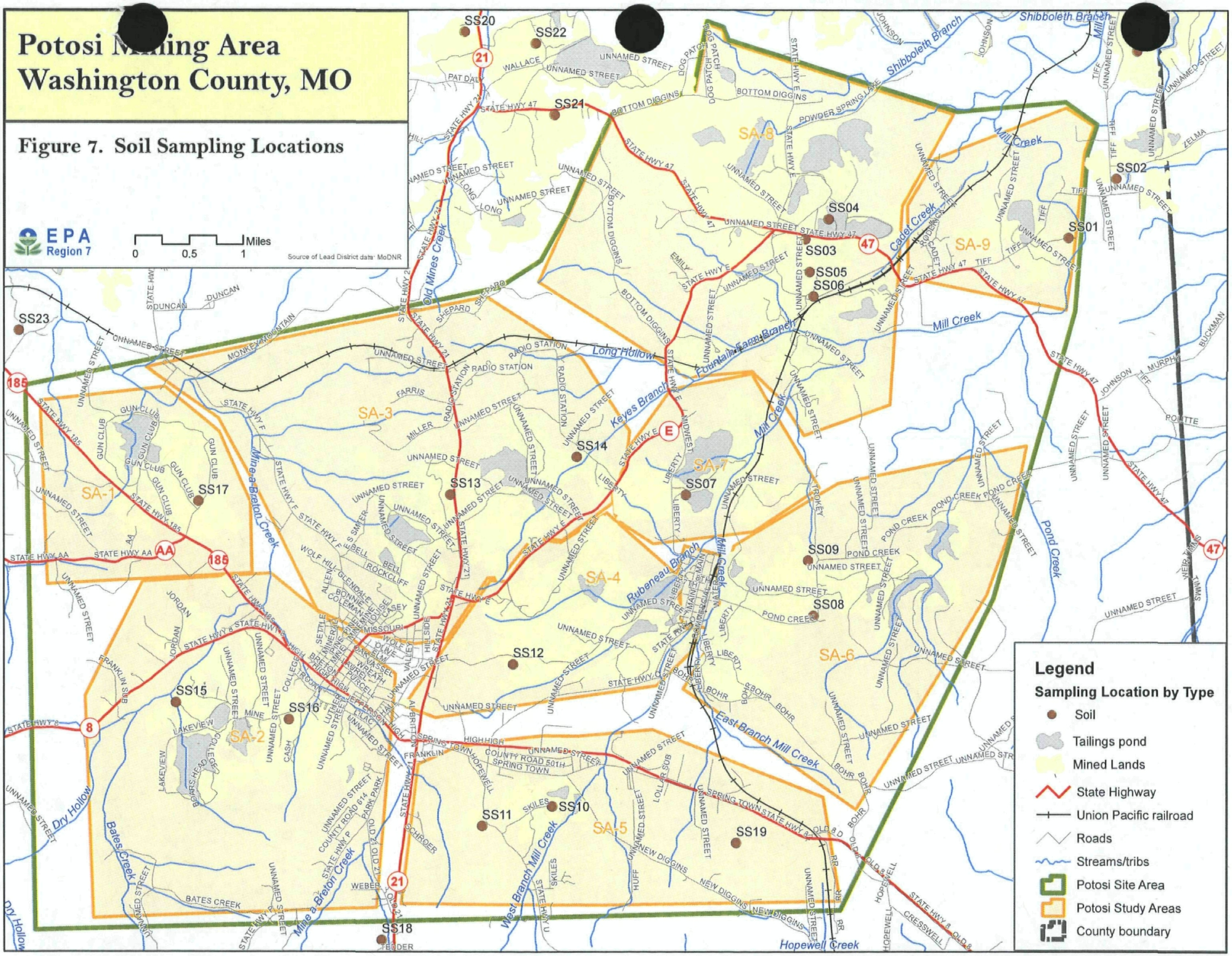
Potosi Mining Area
Washington County, MO

Figure 6. Surface Water and Sediment Sampling Locations



Potosi Mining Area
Washington County, MO

Figure 7. Soil Sampling Locations



APPENDIX C
STANDARD OPERATING PROCEDURES

REAC
Response Engineering and Analytical Contract

STANDARD OPERATING PROCEDURES

BENTHIC MACROINVERTEBRATE SAMPLING

CONTENTS

CONTENTS (cont'd)

1.0 SCOPE AND APPLICATION

The purpose of this Standard Operating Procedure (SOP) is to describe the procedures for sampling benthic macroinvertebrate populations. Analysis of benthos will be used, in conjunction with other bioassessment techniques, to assess the direct and/or indirect impact of contamination to benthic communities, composition, and functionality.

2.0 METHOD SUMMARY

Benthic sampling can be qualitative (a general assessment of the benthic taxa present, possibly with some observations of their relative abundance) or quantitative (an estimate of the numbers [total or by taxa] present so that a statistical confidence interval of the estimate can be calculated). Quantitative sampling is necessary to determine ratios of various functional feeding groups of benthic macroinvertebrates; for example, the ratio of the number of forms that skeletonize leaf litter (shredders) to the number that graze on attached algae (scrapers). All representative subhabitats of a given system should be thoroughly sampled. Different methods may be used depending on the type of habitat sampled (e.g., lentic or lotic).

2.1 Lentic Habitats

A variety of bottom dredges (grabs) are available for use in lentic, or still water habitats. Examples of some common collecting instruments are the Ekman dredge, Peterson dredge, and Ponar dredge. The Ekman dredge is the easiest to use since it is light and relatively easy to "set". However, use is limited to soft mud, silt, or finely divided sand bottoms. For sampling where the bottom material is compacted or consists of pebble, gravel, or organic litter substrate, the Peterson or Ponar dredge is preferred since these dredges are heavier than the Ekman dredge. Usually, small obstacles will not prevent the closing of these dredges and are crushed by the jaws, whereas the same materials may block the operation of the Ekman dredge.

2.2 Lotic Habitats

Benthic macroinvertebrate sampling from lotic, or running water habitats differs in both the type of the organisms collected, and the means used for collection. Because of the scouring action of the current, soft sediments are rarely found. Organisms of running waters are usually heavy bodied or have special means of attachment. Finally, because of the lack of fine sediments, the distribution of organisms with depth in the sediment is usually greater in running waters than in standing waters. An example of a commonly utilized sampling device used in running waters is the Surber stream bottom sampler. This device has a major drawback in that it's use is restricted to waters of less than 30 centimeters (cm) in depth and of slow to moderate velocity.

3.0 SAMPLE PRESERVATION, CONTAINERS, HANDLING, AND STORAGE

After a representative sample has been collected, animals, vegetation, and substrate are preserved for picking, sorting, and analysis (e.g., taxonomic, statistical). The organisms from each sample are placed into a separate jar or vial and covered with a preservative such as 70 percent (%) ethyl alcohol, 40% isopropyl alcohol, or neutral formalin. Alcohol is less irritating to use than formalin but should be used only for short-term storage unless the animals are fixed first in formalin. Organisms collected for residue analysis are not chemically preserved, but are frozen on dry ice.

4.0 INTERFERENCES AND POTENTIAL PROBLEMS

There are several potential problems and interferences, most of which are physical in nature and may occur when sampling benthic communities. Temperature, recent storm events, high/low water levels and other factors can impact local benthic communities. Care must be taken to ensure that the sample locations are not impacted by these factors. If it cannot be avoided (e.g., flood event), the problem/interference should be properly documented in field logbooks as per REAC SOP #4001, *Logbook Documentation*.

5.0 EQUIPMENT/APPARATUS

The following equipment is useful for benthic sampling:

- Surber stream bottom sampler
- Trowels
- Forceps
- Stiff bristle brush (wire or nylon)
- Dredge, line, messenger weight, and pole
- Plastic bucket, 5-gallon
- No. 35 (0.5 millimeter) soil sieve
- Wide-mouthed high density polyethylene (HDPE) bottles (500 to 1,000 milliliters)
- D-frame nets
- Hip or chest waders
- Artificial substrates (e.g., Hester and Dendy)
- Anchor for artificial substrates
- White plastic trays or pans
- Wooden stakes
- Flagging tape
- Marking pens
- Shoulder length gloves
- Acetate cores with caps
- Surgical gloves
- Tape measure

6.0 REAGENTS

Buffered neutral formalin is used to fix samples, and 70% ethyl alcohol or 40% isopropyl alcohol are used for sample preservation when samples are archived or submitted for taxonomic analysis only.

7.0 PROCEDURES

7.1 Preparation

1. Determine the extent of the sampling effort, the sampling methods to be employed, the types and amounts of equipment and supplies needed.

2. Ensure that all equipment is in working order.

3. Perform a general survey prior to site entry in accordance with the Work Plan (WP) and Health and Safety Plan (HASP).

4. Use stakes, flagging tape, buoys, or global positioning system (GPS) generated waypoints to identify and mark all sampling locations.

7.2 Surber Stream Bottom Sampler

1. Select a sampling site representative of the area desired with a depth no greater than the height of the net frame, or a depth easily reached by the sampler. Shallow riffle areas of relatively fast moving streams with cobble, gravel, and sand substrates are often best sampled using this method. The velocity of the stream must not be so great as to cause a "pressure head" of water to flow around the mouth of the sampler.

2. Wade from downstream, and place the sampler with the mouth of the net facing upstream in an undisturbed area. Care must be taken that there is no disturbance of the substrate upstream from the net, since organisms may be dislodged and washed into the net.

3. Lower the square foot frame onto the substrate anchoring it in place by setting the sampler between the feet of the person sampling. Pick up the larger rocks or bits of substrate within the frame, and while holding them in the mouth of the net, brush them free of all organisms, allowing the current to carry them into the net. Discard these rocks outside the frame.

4. After all the organisms have been brushed off the larger rocks and debris, use hands or a trowel to stir up or agitate the substrate within the square foot frame so that dislodged organisms can be carried into the mouth of the net and not around it. The substrate within the frame should be disturbed to a uniform depth.

5. After a sample has been collected, empty the contents of the net into a white pan, and remove rocks and large debris after inspection for clinging organisms. When completed, transfer the contents of the white pan into a wide-mouthed jar. Turn the net "wrong side out", and using forceps, pick off any organisms that have attached to the net fabric and place

them into the jar. Depending on field conditions, number of organisms, etc., organisms may be partially or fully picked in the field. This may save time compared to laboratory processing, since the organisms are alive and easier to spot. If required, target organisms being submitted for residue analysis should be removed, placed into an appropriate container, and stored on dry ice.

6. Depending on the number of replicates desired, move the sampler to other sites, and repeat steps 1 through 5.

7. After sampling is complete, add a sufficient amount of fixative or preservative to cover the substrate and organisms in the bottle. Each jar should be labeled indicating the date, site name, sample location and replicate number. Samples should be shipped to the appropriate laboratory for further processing as per REAC SOP #2004, *Sample Packaging and Shipment.*

7.3 Hess Sampler

1. Select a sampling site representative of the area desired with a depth no greater than the height of the sampler, or a depth easily reached by the sampler. Due to the design of the sampler, deeper riffle areas (too deep for use with a Surber Sampler) of relatively fast moving streams with cobble, gravel, and sand substrates are often best sampled using this method.

2. Wade from downstream, and embed the base of the sampler into the stream substrate with the sampler body mesh facing upstream and the sample collection net facing downstream in an undisturbed area.

3. Set the sampler between the feet of the person sampling. Pick up the larger rocks or bits of substrate within the sampler, and while holding them in the mouth of the net, brush them free of all organisms, allowing the current to carry them into the net. Discard these rocks outside the frame.

4. After all the organisms have been brushed off the larger rocks and debris, use hands or a trowel to stir up or agitate the substrate within the sampler so that dislodged organisms can be carried into the mouth of the net. The substrate within the frame should be disturbed to a uniform depth.

5. After a sample has been collected, empty the contents of the net into a white pan, and remove any large debris after inspection for clinging organisms. When completed, transfer the contents of the white pan into a wide-mouthed jar. Turn the net "wrong side out", and using forceps, pick off any organisms that have attached to the net fabric and place them into the jar. Depending on field conditions, number of organisms, etc., organisms may be partially or fully picked in the field. This may save time compared to laboratory processing, since the organisms are alive and easier to spot. If required, target organisms being submitted for

residue analysis should be removed, placed into an appropriate container, and stored on dry ice.

6. Depending on the number of replicates desired, move the sampler to other sites, and repeat steps 1 through 5.

7. After sampling is complete, add a sufficient amount of fixative or preservative to cover the substrate and organisms in the bottle. Each jar should be labeled indicating the date, site name, sample location and replicate number. Samples should be shipped to the appropriate laboratory for further processing as per REAC SOP #2004, *Sample Packaging and Shipment.*

7.4 Dredges

1. Select sampling sites representative of the area to be sampled, and determine the number of replications desired. Dredges are particularly useful in areas of deep water, or soft bottom sediments.

2. Secure the dredge to a length of heavy rope. Ekman dredges can be operated by securing the dredge and a weighted messenger to a length of rope, or by bolting it to the end of the pole. After the selected dredge is secured, set the trip mechanism.

3. Lower the dredge slowly, particularly through the final 0.5 meters (m) of water above the substrate surface. If the depth of the water is unknown, lower the dredge slowly to the bottom and then raise it, move 1 or 2 meters laterally, and re-lower gently. Trip the dredge by dropping the weighted messenger, or depressing the tripping mechanism on the pole for the Ekman, or by allowing the line to slacken for the Peterson dredge or Ponar.

4. Retrieving and emptying the contents of a dredge requires two people due to the weight and bulkiness of a filled dredge. Lift the filled dredge to the surface with a smooth, even motion to avoid jarring out contents. It is recommended that the dredge be held over a pail while it is brought over the side of a boat or recovered. Care must be taken when retrieving and handling a dredge since they are especially heavy when loaded.

5. Empty the contents of the dredge into a pail. Repeat the sampling procedure until all of the desired replicates (each replicate is considered one discrete sample) have been collected at the sampling location.

6. Gently pour the contents of the pail into a No. 35 soil sieve. Repeat this step until all of the contents of the pail have been poured through the sieve. A gentle spray of water can be used to break up compacted particles, and to facilitate passing the sediment through the sieve.

7. The material remaining on the sieve is the collected sample. Place the sieve into a pail of water, or surface water being sampled and agitate, taking care not to let the water run over the top of the sieve. This will wash smaller material through the sieve, and will concentrate the remaining material at one edge if the sieve is held at an angle.

8. When all materials and organisms are concentrated on one side of the sieve, remove the rocks and large debris that are free of clinging organisms and scrape them into a wide-mouthed jar. If necessary, wash the sieve again, and place any additional organisms/materials left in the sieve into the bottle, picking out the remains with forceps if necessary.

9. After sampling is complete, add a sufficient amount of fixative or preservative to cover the substrate and organisms in the bottle. Each jar should be labeled indicating the date, site name, sample location and replicate number. Samples should be shipped to the appropriate laboratory for further processing as per REAC SOP #2004, *Sample Packaging and Shipment*.

7.5 Kick Nets

1. Select sampling sites representative of the area to be sampled, and determine the number of replicates desired. Riffle areas of relatively fast moving streams with cobble, gravel, and sand substrates are often best sampled using this method. This sampling method requires at least two people. The name "kick net" describes its functionality where, a field team member kicks up the substrate upstream, and allows the material to flow into the net.

2. The first field team member wades in from downstream, and places the net with the mouth facing upstream in an undisturbed area. The net is gently placed on the stream bottom in a relatively flat area and not on large rocks or debris.

3. While one field team member holds the net in place, another carefully wades into the stream perpendicular to the current, approximately 1 to 2 meters (m) upstream of the net. This team member begins kicking up the stream bottom while moving steadily downstream toward the net. This method can be "area" specific, or "timed". Once the area has been disturbed throughly, the person holding the net lifts the net out of the water with a gentle scooping motion arcing the net forward and upward.

4. After a sample has been collected, empty the contents of the net into a white pan, and remove any debris after inspection for clinging organisms. When completed, transfer the contents of the white pan into a wide-mouthed jar. Turn the net "wrong side out", and using forceps, pick off any organisms that have attached to the net fabric and place them into the jar. Depending on field conditions, number of organisms, etc., organisms present may be partially or fully picked in the field. This may save time compared to laboratory processing since the organisms are alive and easier to spot. If required, target organisms being

submitted for residue analysis should be picked out and placed into the appropriate container and stored on dry ice.

5. Depending on the number of replicates desired, move the sampler to other sites, and repeat steps 1 through 4.

6. After sampling is complete, add a sufficient amount of fixative or preservative to cover the substrate and organisms in the bottle. Each jar should be labeled indicating the date, site name, sample location and replicate number. Samples should be shipped to the appropriate laboratory for further processing as per REAC SOP #2004, *Sample Packaging and Shipment.*

7.6 Artificial Substrates

1. Select sampling sites representative of the area to be sampled, and determine the number of replicates desired. Artificial substrates are particularly useful in areas of deep water, swift currents, or soft bottom sediments.

2. Anchor the substrates to a weighted object. The anchor and the means of attachment will be dependent on existing conditions at the sampling location. For example, the threaded "eye" bolt used to assemble a Hester and Dendy may be threaded onto a steel frame.

3. Place the substrates in the area to be sampled, and allow them to remain in place for an appropriate period of time (typically one month) in order for macroinvertebrates to colonize.

4. After the pre-determined time, retrieve the substrates and place into a jar to be preserved, or scrape off the colonists using a soft rubber spatula and place into a wide-mouth jar. If required, target organisms being submitted for residue analysis should be picked out and placed into the appropriate container and stored on dry ice.

5. After sampling is complete, add a sufficient amount of fixative or preservative to cover the substrate and organisms in the bottle. Each jar should be labeled indicating the date, site name, sample location and replicate number. Samples should be shipped to the appropriate laboratory for further processing as per REAC SOP #2004, *Sample Packaging and Shipment.*

7.7 Sediment Cores

1. Select sampling sites representative of the area to be sampled, and determine the number of replicates desired. Sediment coring is particularly useful in estuarine environments such as tidal flats or salt marshes, and is often conducted during low tide. It is suggested that the cores be no less than 8.8 centimeters (cm) in diameter, and 10 cm in length.

2. Push the core into the sediment to the desired depth. Remove the core from the sediment by

capping the top of the core and by gently rotating it as it is slowly withdrawn from the sediment. Once removed, cap the bottom end of the core for later processing. Repeat until all replicates have been taken.

3. Gently pour the contents of the core into a No. 35 soil sieve. The core may be rinsed with water to remove any sediment adhering to the insides of the core. After the core is emptied a gentle spray of water can be used to break up compacted particles, and to facilitate passing the sediment through the sieve.

4. The material remaining on the sieve is the collected sample. Place the sieve into a pail of water, or surface water being sampled and agitate, taking care not to let the water run over the top of the sieve. This will wash smaller materials through the sieve, and will concentrate the materials at one edge if the sieve is held at an angle.

5. When all of the materials and organisms are concentrated on one side of the sieve, scrape them into a wide-mouthed jar. If necessary, wash the screen again, and place any additional organisms/materials left in the sieve into the bottle, picking out any remains with forceps if necessary.

6. After sampling is complete, add a sufficient amount of fixative or preservative to cover the substrate and organisms in the bottle. Each jar should be labeled indicating the date, site name, sample location and replicate number. Samples should be shipped to the appropriate laboratory for further processing as per REAC SOP #2004, *Sample Packaging and Shipment.*

7.8 Post Operation

7.8.1 Field

Following all sampling events any equipment used should be cleaned, and if necessary decontaminated as per Environmental Response Team (ERT)/REAC SOP #2006, *Sample Equipment Decontamination.*

7.8.2 Laboratory Processing

The preserving fluid may be decanted off and the sample rinsed with water to prevent exposure of preservative fumes to the processor. Empty the contents of a sample container into a No. 35 sieve and rinse with water to remove the fixative or preservative. The rinsate should be collected and disposed of properly. Empty the contents of the sieve onto a white plastic tray. Samples collected may contain a mixture of mud, rocks, sand, and debris, in addition to the desired organisms. Remove the organisms from the unwanted material and separate them into similar taxonomic groupings (e.g., Order; Family) for identification and enumeration. It is recommended that the processor use illuminated dissecting lamps to aid in this task.

One benthos sample may have an excessively high density of organisms which could require several person-hours for processing. In such a case, a sub-sample may be collected from that sample. For example, the bottom of the white tray is separated into a grid consisting of 100 numbered squares. Empty the contents of the rinsed sieve, and evenly spread out the sample to cover the grid. Using a random number generator, ten squares are selected and the organisms within the squares are removed. This is the representative sample for that particular location/replicate. A variety of laboratory techniques, each specifically suited for a particular type of sample, are used by different investigators. Techniques should be modified where appropriate. The processor should be aware of whatever procedure is employed so that the final count of organisms is representative of those found in the habitat sampled.

7.8.3 Office

All field notes and/or other logging information should be included in the final report following the appropriate format outlined in REAC SOP #4021, *Preparation of Final Reports*.

8.0 CALCULATIONS

Various indices may be calculated using the results of the final count of organisms. Calculations utilized may be project specific and may include, but are not limited to: total number of organisms, total number of taxa, ratio of pollution sensitive organisms to pollution tolerant organisms (e.g., Ephemeroptera, Plecoptera, Trichoptera [EPT]:chironomid ratio), percent dominant taxa, diversity indices, and statistical analysis.

9.0 QUALITY ASSURANCE/QUALITY CONTROL

The following Quality Assurance/Quality Control (QA/QC) procedures apply:

1. All samples must be documented on chain of custody forms, field data sheets or in site logbooks as per REAC SOPs #4005 *Chain of Custody Procedures*, #4001 *Logbook Documentation,* and #2002 *Sample Documentation.*

2. Prior to sampling, the number and size of samples will be outlined in the site-specific WP.

3. All deliverables will receive a peer review prior to release as per REAC Administrative Procedure (AP) #22, *Peer Review of REAC Deliverables*.

10.0 DATA VALIDATION

Taxonomic information will be confirmed by an experienced biologist familiar with benthic organisms. Thoroughness of the sorting will be verified by having an additional person re-process ten percent of the samples. If it is determined that a large number of organisms were overlooked ,then all the samples will be re-

processed.

11.0 HEALTH AND SAFETY

When working with potentially hazardous materials, follow U.S. EPA, Occupational, Safety, and Health Administration (OSHA), and corporate health and safety procedures. More specifically, refer to REAC SOP #3001, *REAC Health and Safety Program Policy and Implementation.*

12.0 REFERENCES

Although not cited in this document, the following documents have been useful, and are recommended references to the methods discussed in this SOP.

Brower, J.E. and J.H. Zar, 1984. Field and Laboratory Methods for General Ecology. William C. Brown Publishers. Dubuque, Iowa.

Lind, Owen T., 1979. Handbook of Common Methods in Limnology, Second Edison. C.V. Mosby Company. St. Louis, Missouri.

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U.S. EPA. 1990. *Macroinvertebrate Field and Laboratory Methods for Evaluating the Biological Integrity of Surface Waters.* U.S. Environmental Protection Agency, Office of Research and Development, Washington, DC. EPA/600/4-90/030.

U.S. EPA. 1999. *Rapid Bioassessment Protocols for Use in Streams and Wadeable Rivers: Periphyton, Benthic Macroinvertebrates, and Fish, Second Edition.* U.S. Environmental Protection Agency, Office of Water, Washington, DC. EPA 841-B-99-002.

U.S.G.S. 2000. *Methods of Analysis by the U.S. Geological Survey National Water Quality Laboratory-Processing, Taxonomy, and Quality Control of Benthic Macroinvertebrate Sample.* U.S. Geological Survey Chief, National Water Quality Laboratory, Denver, CO. Open File Report 00-212.

13.0 APPENDIX

This section is not applicable to this SOP.

U. S. EPA ENVIRONMENTAL RESPONSE TEAM

STANDARD OPERATING PROCEDURES

TERRESTRIAL PLANT COMMUNITY SAMPLING

CONTENTS

SUPERCEDES: SOP #2037; Revision 0.0; 01/24/92; U.S. EPA Contract 68-03-3482.

TERRESTRIAL PLANT COMMUNITY SAMPLING

1.0 SCOPE AND APPLICATION

This standard operating procedure (SOP) describes the method for sampling terrestrial plant communities on hazardous waste sites. Analysis of vegetation will be used, in conjunction with other bioassessment techniques, to assess the impact of site contamination on plant life. Vegetation will be evaluated for shifts in community structure as a function of site contamination. Included below are procedures for obtaining representative measurements and guidance on quality assurance/quality control measures.

These are standard (i.e., typically applicable) operating procedures which may be varied or changed as required, dependent upon site conditions, equipment limitations, or limitations imposed by the procedure. In all instances, the ultimate procedures employed should be documented and associated with the final report.

Mention of trade names or commercial products does not constitute U.S. Environmental Protection Agency (U.S. EPA) endorsement or recommendation for use.

2.0 METHOD SUMMARY

The use of this SOP is dependent on weather and season. Non-woody plants will not endure throughout a winter with freezing temperatures, and thus cannot be evaluated by these methods during this part of the year in such climates.

A survey of site history will be made with all readily available information. Information on site contaminants, site and regional vegetation, and local climatic conditions will be considered. Remote sensing and topographic maps, when available, will be obtained and reviewed. Information on rare and endangered flora that may exist within the study areas should be obtained and reviewed.

Plots and transects are used to collect information representative of vegetative communities of the study site. Choice of appropriate sampling technique (i.e., plots vs. transects) depends upon site characteristics, plant characteristics, and study objectives. Information concerning species identification, enumeration, spatial arrangement, and size/shape attributes of the vegetation will be recorded in logbooks and on field data sheets. Signs of stressed vegetation will be noted. Samples representative of study location flora will be gathered for taxonomic verification. Values for species density, coverage, and frequency will be computed, as necessary.

3.0 SAMPLE PRESERVATION, CONTAINERS, HANDLING, AND STORAGE

Samples of vegetation may be required for taxonomic verification. Whole plants or selected parts (i.e., leaves, twigs, or flowers) will be placed in a resealable plastic bag and kept cool (4°C) to slow decay. All materials, with the exception of woody specimens, should be kept from temperature extremes and should be identified as soon as possible. If more than a week will pass before the samples can be identified, the samples will be placed in a plant press. Samples may also be archived by placing them in a plant press after identification.

4.0 INTERFERENCES AND POTENTIAL PROBLEMS

There are several potential problems and interferences that may occur when sampling plant communities.

1. Access to study locations must be obtained prior to study commencement.

2. Environmental disturbances, such as drought or fire, may confound data collection and interpretation. In addition, physical disturbances by man, such as the mowing or trampling of site vegetation, will further complicate assessment.

3. Microclimatic differences, such as sun/shade and moisture/drought, will affect plant growth and response.

5.0 EQUIPMENT/APPARATUS

Equipment needed for plant community sampling may include, depending upon the study objectives, the following items:

- Stakes - with sufficient height to be observed and sufficient width to stay in place during the period of study
- Line or rope
- Tape measure and/or plot frames
- Shovels and hand trowels - both of which must have unpainted stainless steel blades
- Pruning shears and/or knives
- Resealable plastic bags
- Cooler with ice
- Regional field guides to native plants
- Compass
- Vernier calipers
- Clinometer (optional) - necessary when measuring tree heights
- Documentation supplies (includes logbook, chain of custody records and custody seals, field data sheets and sample labels)
- Plant press (optional)

6.0 REAGENTS

Reagents are not required for preservation of vegetation samples. Samples should, however, be cooled to 4°C in order to minimize the degree of deterioration. Decontamination of sampling equipment may be required. Decontamination solutions are specified in ERT/REAC SOP #2006, Sampling Equipment Decontamination.

7.0 PROCEDURES

7.1 Sampling Considerations

7.1.1 General Site Survey

Prior to initiation of vegetation sampling, the appropriate sample collection area(s) should be determined. This may be accomplished with the assistance of remote sensing and/or topographic maps. Field guides to the regional vegetation species and experts knowledgeable about local conditions should be consulted. The extent of contamination should be established.

Consideration must also be given to the location of specific sampling points so that they provide representative samples (Section 7.1.2). The presence of rare or endangered species should also be determined and care taken not to adversely impact these communities during site activities.

A site sampling plan which details the number and general areas to be assessed will be prepared prior to plant community sampling activities.

7.1.2 Representative Samples

For representative sample collection, seasonal community fluctuations should be determined and climatic patterns analyzed. Topography and soil types should also be considered.

Sampling of vegetation should occur during seasons of the year where the species of interest are present. For example, if a complete vegetation survey were to be performed, plant assessment may be required over several seasons. If the species of concern were annuals, vegetation study should occur during the growing season while these species display characteristics that can be observed. Additionally, depending upon the study objectives, it may be necessary to survey plant communities several times during the growing season or throughout the year.

7.2 Sample Collection

The ecological parameters of density, coverage, and frequency reflect vegetational community structure and are those that are discussed in this SOP. Additional information may be collected for use in studies of plant community structure. Additional parameters useful in determining and comparing plant community structure include diversity and similarity indices. These parameters will not be addressed in the present SOP; however, measurements used to calculate these parameters may be collected at the same time as sampling activities described in this SOP. For a description of these additional parameters, refer to Brower and Zar.[1]

The size, shape, and number of vegetation sample locations ultimately depends upon the vegetation type present (i.e., herb, shrub, tree, vine, etc.) and their distribution pattern. Basically, there are two general approaches to plant community sampling: plots/quadrats and transects.

7.2.1 Sample Plots/Quadrats

A sample plot or quadrat is the specific area within which vegetation analysis will occur. The number, size, shape, and location of sample plots will depend upon the types of vegetation to be sampled and the objectives of the study. For example, smaller plots may be required for a site with dense or rich flora.

Typically, rectangular or circular plots are used. Circular plots are easy to set up. They require only a stake and premeasured line (or measuring tape). Circular plots are often used in the assessment of woody species. However, rectangular plots have been found, in general, to yield better results for plant surveys.[1] Rectangular plots require at least four stakes and a plot frame of desired size (or measuring tape and a means to make right angles) to be constructed.

The following procedure will be followed when surveying plant communities:

1. Divide vegetational areas of the site to be assessed into a grid. If soil/sediment sampling is also performed, it is most efficient and advantageous to use the same sample location grid for both soil/sediment sampling and plant community assessment. When vegetation is collected for analysis, use of the same grid locations will provide the potential for comparison of contaminant concentrations in the soil/sediment and the vegetation.

2. Select locations for a predetermined number of plots (as described in the site sampling plan) using randomly-selected grid coordinates. (X and Y coordinates can simply be paced out from the appropriate axis.)

3. Establish plots according to study objectives and the following vegetation classifications:

   a. Closely Spaced Herbs - [plants of less than 1 meter (m) in height]
   - use a rectangular plot of 1 m^2 (for example, 1.0 m x 1.0 m)

   b. Bushes/Saplings/Shrubs - [woody plants with height greater than 1 m and main stem diameter of less than 10 centimeters (cm), excluding vines]
   - use a plot area of 10 m^2 (for example, 2.5 m x 4.0 m)

   c. Trees - [any non-climbing woody plants with main stem diameter at breast height (DBH) of greater or equal to 10 cm. (DBH = 1.5 m above ground level)]
   - identify each tree within a 10 meter radius of the selected center point of the sample plot

d. Woody Vines (Lianas) - (woody climber with DBH of less than 10 cm)
- identify each vine within a 10 meter radius of the selected center point of the sample plot (usually associated with tree plots)

4. Identify and count species in each plot.

5. Estimate species coverage within plot area. Measure DBH for tree species, when applicable, to calculate basal area form which cover estimates are made.

6. Note visual cues of stress and overall health of plot vegetation (including wilting, browning, stunted growth, chlorosis, etc.).

7. Note habitat characteristics (for example, moisture availability, degree and direction of exposure of slope, tidal location, etc.).

8. Collect vegetative samples from each plot, as necessary, for taxonomic verification. Store samples as described in Section 3.0.

9. Repeat the above procedures for an uncontaminated reference area during the same period of study.

10. Perform appropriate calculations (Section 8.0) and appropriate statistical analyses upon the data.

11. Prepare generalized vegetation map showing plant communities and sampling locations.

7.2.2 Transect Sampling

When the use of plots is impractical, transects may be used. Transects are especially useful in the evaluation of transitional communities. Ecological parameters that are studied utilizing plots can be studied utilizing transects. Additionally, changes in the vegetation in relation to environmental gradients may be observed. The type, size, number, and locations of transects chosen will depend upon study objectives, vegetation type, and site characteristics. Longer transects should be made when plants are widely dispersed.

Types of transects include belt transects and line intercept transects. A belt transect is a line transect with width. It is essentially a long, thin quadrat or can be divided into zones (each of which act as plots). In the line intercept method a known length of rope or tape measure is laid out in a line and information is collected as vegetation intercepts the line. The line intercept method is particularly useful for surveys of shrubs. This method is used for vegetative cover estimates and species composition estimates. With this method, only estimates of linear density can be made, as area is not involved.

TERRESTRIAL PLANT COMMUNITY SAMPLING

The following procedure applies to plant community sampling using transects:

1. Determine which transect method best suits the objective(s) of the study and habitat available.

2. Establish transects according to the study objectives and the appropriate transect method:

   a. Belt transect

      - establish transect length and width
      - locate belt transect(s) randomly in the selected study area(s) or with bias along a specific gradient or feature of interest
      - identify and count species
      - estimate coverage and measure DBH (on woody species, when required) within plot(s)

   b. Line intercept

      - establish transect length
        Short lines (under 50 m) are used for assessment of herb species
        Long lines (greater then 50 m) are used for assessment of some shrub and tree communities
      - locate transect line(s) randomly in the selected study area(s) or with bias along a specific gradient or feature of interest
      - divide transect line into equal intervals
      - record the length of the line intercepted for each plant intercepting the line
      - count, measure, and identify plants that either intercept the transect line or are within a small distance from the line, depending upon the density of the vegetation

3. Note visual cues of stress and overall health of plot vegetation (including wilting, browning, stunted growth, chlorosis, etc.).

4. Note habitat characteristics (for example, moisture availability, degree and direction of exposure of slope, tidal location, etc.).

5. Collect vegetative samples from each transect, as necessary, for taxonomic verification. Store samples as described in Section 3.0.

6. Repeat the above procedures for an uncontaminated reference area during the same period of study.

7. Perform appropriate calculations (Section 8.0) and appropriate statistical analyses upon data.

8. Prepare a generalized vegetation map showing plant communities and sampling locations.

7.3 Sample Collection Variation

Taxonomic identification to the species level is often required for the vegetation assessment methods described. When no such knowledge is desired and/or available, a generalized physiognomic approach may be utilized. Physiognomy is the study of form, structure, and spatial arrangement of an organism. The resulting data may be sufficiently detailed and organized and can be collected comparatively rapidly.

Physiognomic characteristics that may be observed and documented include:

- Life form - presence, dominance, or absence of specific structural life forms (herbs, trees, vines, etc.)
- Stratification and zonation - layers of vegetation from the ground-layer to the canopy
- Foliage density - amount of shading vs. light penetration
- Coverage - sparse (less than five percent coverage) to dense (greater than 75% coverage)
- Dispersal pattern - arrangement of species (rows, clumps, solitary, etc.)
  - uniformity (evenly-spaced vs. irregularly distributed)
  - spacial separation (distant vs. dense)

8.0 CALCULATIONS

8.1 Calculations for Plots and Belt Transects

Density for Species i (D_i)

$$D_i = n_i/A$$

Where:
n_i = total individuals for species i
A = total area sampled

Relative Density for Species i (RD_i)

$$RD_i = n_i/\Sigma n$$

Where:
n_i = number of individuals of species i
Σn = total number of individuals of all species in sampled plots

Coverage for Species i (C_i)

$$C_i = a_i/A$$

Where:
a_i = total area covered for species i
A = total area sampled

Relative Coverage of Species i (RC_i)

$RC_i = C_i/\Sigma C$

Where:
C_i = coverage for species i
ΣC = sum of coverage for all species

Frequency of Species i (f_i)

$f_i = j_i/k$

Where:
j_i = number of plots containing species i
k = total number of plots

Relative Frequency of Species i (RF_i)

$RF_i = f_i/\Sigma f$

Where:
f_i = frequency of species i
Σf = sum of frequencies of all species

8.2 Calculations for Line Transects

Linear Density Index of Species i (ID_i)

$ID_i = n_i/L$

Where:
n_i = number of individual of species i
L = total length of all sampled transects

Relative Density for Species i (RD_i)

$RD_i = n_i/\Sigma n$

Where:
n_i = number of individual of species i
Σn = total number individuals of all species in sampled transects

Linear Coverage Index of Species i (IC_i)

$IC_i = l_i/L$

Where:
l_i = sum of intercept lengths intercepted by species i
L = total length of all sampled transects

Relative Coverage of Species i (RC_i)

$RC_i = l_i/\Sigma l$

Where:
l_i = sum of intercept lengths intercepted by species i
Σl = sum of intercept lengths for all species intercepting transects

Frequency of Species i (f_i)

$f_i = j_i/k$

Where:
j_i = number of intervals containing species i
k = total number of intervals on transects

Relative Frequency of Species i (RF_i)

$RF_i = f_i/\Sigma f$

Where:
f_i = frequency of species i
Σf = sum of frequencies of all species

8.3 Additional Calculation for Tree Species

Basal Area at Breast Height (A), calculated for each tree

$A = pi(r^2)$

Where:
pi = 3.1416
r = radius (in cm)

9.0 QUALITY ASSURANCE/QUALITY CONTROL

The following quality assurance/quality control procedures apply:

1. All data must be documented on field data sheets or within field/site logbooks.
2. All instrumentation must be operated in accordance with the operating instructions as supplied by the manufacturer, unless otherwise specified in the work plan. Equipment checkout and calibration activities must occur prior to sampling/operation and they must be documented.
3. Calculations will be checked by an additional person at a rate of ten percent.
4. A sampling plan, including sample size, will be created prior to sampling.

10.0 DATA VALIDATION

Data generated will be reviewed according to the quality assurance/quality control considerations listed in Section 9.0.

In addition, taxonomic information will be confirmed by a regional biologist familiar with the site's vegetation.

11.0 HEALTH AND SAFETY

When working with potential hazardous materials, follow U.S. EPA, OSHA, and corporate health and safety procedures.

When sampling at a known or suspected contaminated site, precautions must be taken to safeguard the samplers from chemical and physical hazards. In addition, it would benefit the samplers to be familiar with and avoid any contact with plants that present a contact hazard such as poison ivy, poison sumac, and poison oak.

12.0 REFERENCES

[1]Brower, J.E., and J.H. Zar, "Field and laboratory methods for general ecology," William C. Brown Publishers, Dubuque, Iowa, 1984.

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REAC
Response Engineering and Analytical Contract

STANDARD OPERATING PROCEDURES

SEDIMENT SAMPLING

CONTENTS

9.0 QUALITY ASSURANCE/QUALITY CONTROL*

CONTENTS (Cont'd)

* These sections are affected by Revision 0.0.

SUPERSEDES: SOP #2016; Revision 0.0; 11/17/94; U.S. EPA Contract 68-C4-0022.

1.0 SCOPE AND APPLICATION

This Standard Operating Procedure (SOP) is applicable to the collection of representative sediment samples. Analysis of sediment may be biological, chemical, or physical in nature and may be used to determine the following:

- toxicity;
- biological availability and effects of contaminants;
- benthic biota;
- extent and magnitude of contamination;
- contaminant migration pathways and source;
- fate of contaminants;
- grain size distribution;
- deposition environment;
- sediment type.

For the purpose of this procedure, sediment is the mineral and/or organic material situated beneath an aqueous layer. The aqueous layer may be either static, as in lakes, ponds, and impoundments or flowing, as in rivers and streams. The methodologies discussed in this SOP are applicable to the sampling of sediment in both flowing and standing water.

These are standard (i.e. typically applicable) operating procedures which may be varied or changed as required, dependent upon site conditions, equipment limitations or limitations imposed by the procedure. In all instances, the ultimate procedures employed should be documented and associated with the final report.

Mention of trade names or commercial products does not constitute United States Environmental Protection Agency (U.S. EPA) endorsement or recommendation for use.

2.0 METHOD SUMMARY

Sediment samples may be collected using a variety of methods and equipment, depending on the depth of the aqueous layer, the portion of the sediment profile required (surface vs. subsurface), the type of sample required (disturbed vs. undisturbed), contaminants present, sediment type, and analyses required.

Sediment is collected from beneath an aqueous layer either directly, using a hand-held device such as a shovel, trowel, or auger, or indirectly, using a remotely activated device such as an Ekman or Ponar dredge. Following collection, sediment is transferred from the sampling device to a sample containers of appropriate size and construction for the analysis (es) requested. If composite sampling techniques are employed, multiple grabs are placed into a container constructed of an inert material (e.g. stainless steel), homogenized, and transferred to the sample container(s) appropriate for the analysis (es) requested. The homogenization procedure should not be used if the sample analysis includes volatile organic compounds (VOCs). In this case, sediment, or

multiple grabs of sediment, should be transferred directly from the sample collection device or homogenization container to the sample container. Cores may also be collected directly into an acetate sleeve that serves as the sample container for undisturbed samples.

3.0 SAMPLE PRESERVATION, CONTAINERS, HANDLING, AND STORAGE

1. Chemical preservation of solids is generally not recommended. Cooling to 4 degrees Celsius (°C) is usually the best approach, supplemented by the appropriate holding time for the analyses requested.

2. Wide-mouth glass containers with Teflon™-lined caps are utilized for sediment samples. The sample volume is a function of the analytical requirements and will be specified in the Work Plan or Sampling and Analysis Plan.

3. If analysis of sediment from a discrete depth or location is desired, sediment is transferred directly from the sampling device to a labeled sample container(s) of appropriate size and construction for the analysis (es) requested. Transfer is accomplished with a stainless steel or plastic lab spoon or equivalent.

4. If composite sampling techniques or multiple grabs are employed, equal portions of sediment from each location or collocation are deposited into a decontaminated stainless steel, plastic, or other appropriate container (e.g., Teflon). The sediment is homogenized thoroughly to obtain a mixture representative of the area sampled. The composite sediment sample is transferred to a labeled container(s) of appropriate size and construction for the analysis(es) requested. Transfer of sediment is accomplished with a stainless steel or plastic lab spoon or equivalent. Samples for VOC analysis must be transferred directly from the sample collection device or pooled from multiple areas in the homogenization container prior to mixing. This is done to minimize the loss of contaminant due to volatilization during homogenization.

5. All sampling devices should be decontaminated, then wrapped in aluminum foil. The sampling device should remain wrapped until needed. Dedicated sampling devices should be used for each sample. Disposable sampling devices for sediment are generally impractical due to cost and the large number of sediment samples which may be required. Sampling devices should be cleaned in the field using the decontamination procedure described in Environmental Response Team/Response Engineering and Analytical Contract (ERT/REAC) SOP #2006, *Sampling Equipment Decontamination*.

4.0 INTERFERENCES AND POTENTIAL PROBLEMS

Substrate particle size and organic matter content are a direct consequence of the physical characteristics of a water body and the watershed. Contaminants are more likely to be concentrated in sediment typified by fine particle size and high organic matter. This type of sediment is most likely to be collected from depositional zones. In contrast, coarse sediment with low organic matter does not typically concentrate contaminants and

are generally found in erosional zones. The selection of a sampling location can, therefore, greatly influence the analytical results and should be justified and discussed in the Work Plan or Sampling and Analysis Plan.

5.0 EQUIPMENT/APPARATUS

Equipment needed for collection of sediment samples may include:

- Maps/plot plan
- Safety equipment
- Compass
- Global positioning system (GPS)
- Tape measure
- Survey stakes, flags, or buoys and anchors
- Camera and film
- Stainless steel, plastic, or other appropriate composition bucket
- 4-oz., 8-oz., and one-quart wide mouth jars w/Teflon lined lids
- Ziploc® plastic bags
- Logbook
- Sample jar labels
- Chain of Custody records, field data sheets
- Cooler(s)
- Ice
- Decontamination supplies/equipment
- Spade or shovel
- Spatula
- Scoop
- Trowel (plastic or stainless steel)
- Bucket auger
- Tube auger
- Extension rods and pipe wrenches
- "T" handle
- Sediment coring device (tube, drive head, eggshell check value, nosecone, acetate tube, extension rods)
- Ponar dredge
- Ekman dredge
- Nylon rope or steel cable
- Messenger device
- VibraCore
- Power drill
- S.C.U.B.A. and/or other appropriate dive gear

6.0 REAGENTS

Reagents are not used for preservation of sediment samples. Decontamination solutions are specified in ERT/REAC SOP #2006, *Sampling Equipment Decontamination.*

7.0 PROCEDURES

7.1 Preparation

1. Determine the objective(s) and extent of the sampling effort. The sampling methods to be employed, and the types and amounts of equipment and supplies required will be a function of site characteristics and objectives of the study.

2. Obtain the necessary sampling and monitoring equipment.

3. Prepare schedules, and coordinate with staff, client, and regulatory agencies, if appropriate.

4. Decontaminate or preclean equipment, and ensure that it is in working order.

5. Perform a general site survey prior to site entry in accordance with the site specific Health and Safety Plan (HASP).

6. Use stakes, flags, or buoys to identify and mark all sampling locations. Site specific factors including flow regime, basin morphology, sediment characteristics, depth of overlying aqueous layer, contaminant source, and extent and nature of contamination should be considered when selecting sample locations. If required, the proposed locations may be adjusted based on site access, property boundaries, and surface obstructions.

7.2 Sample Collection

Selection of a sampling device is most often contingent upon: (1) the depth of water at the sampling location, (2) the physical characteristics of the sediment to be sampled, (3) the type of sample required and (4) the parameters being analyzed.

7.2.1 Sampling Surface Sediment with a Trowel or Scoop from Beneath a Shallow Aqueous Layer

For the purpose of this procedure, surface sediment is considered to range from 0 to 6 inches in depth and a shallow aqueous layer is considered to range from 0 to 12 inches in depth. Collection of surface sediment from beneath a shallow aqueous layer can be accomplished with tools such as spades, shovels, trowels, and scoops. Although this method can be used to collect both unconsolidated and/or consolidated sediment, it is limited somewhat by the

depth and movement of the aqueous layer. Deep and rapidly flowing water render this method less accurate than others discussed below. However, representative samples can be collected with this procedure in shallow sluggish water provided care is demonstrated by the sampler. A stainless steel or plastic sampling implement will suffice in most applications. Care should be exercised to avoid the use of devices plated with chrome or other materials; plating is particularly common with garden trowels.

The following procedure will be used to collect sediment with a scoop, shovel, or trowel:

1. Using a decontaminated sampling implement, remove the desired thickness and volume of sediment from the sampling area carefully to minimize movement between sample sediment and water.

2. Transfer the sample into an appropriate sample or homogenization container. Ensure that non-dedicated containers have been adequately decontaminated.

3. Surface water should be decanted from the sample or homogenization container prior to sealing or transfer; care should be taken to retain the fine sediment fraction during this procedure.

7.2.2 Sampling Surface Sediment with a Bucket Auger or Tube Auger from Beneath a Shallow Aqueous Layer

For the purpose of this procedure, surface sediment is considered to range from 0 to 6 inches in depth and a shallow aqueous layer from 0 to 24 inches in depth. Collection of surface sediment from beneath a shallow aqueous layer can be accomplished with a system consisting of a bucket or tube auger, a series of extensions, and a "T" handle (Figure 1, Appendix A). The use of additional extensions in conjunction with a bucket auger can increase the depth of water from which sediment can be collected from 24 inches to 10 feet or more. However, sample handling and manipulation increases in difficulty with increasing depth of water. The bucket or tube auger is driven into the sediment and used to extract a core. The various depths represented by the core are homogenized or a subsample of the core is taken from the appropriate depth.

The following procedure will be used to collect sediment samples with a bucket or tube auger:

1. If the study objectives and characteristics of the sediment or water body warrant, an acetate core may be inserted into the bucket or tube auger prior to sampling. By using this technique, an intact core can be extracted.

2. Attach the auger head to the required length of extensions, then attach the "T" handle to the upper extension.

3. If possible, clear the area to be sampled of any rocks or surface debris.

4. Insert the bucket or tube auger into the sediment at a 0 degrees (°) to 20° angle from vertical. This orientation minimizes spillage of the sample from the sampler upon extraction from the sediment and water.

5. Rotate the auger to cut a core of sediment.

6. Slowly withdraw the auger; if using a tube auger, make sure that the open slot is facing upward.

7. Transfer the sediment into an appropriate sample or homogenization container. Ensure that non-dedicated containers have been adequately decontaminated.

7.2.3 Sampling Deep Sediment with a Bucket Auger or Tube Auger from Beneath a Shallow Aqueous Layer

For the purpose of this procedure, deep sediment is considered to range from 6 to greater than 18 inches in depth and a shallow aqueous layer from 0 to 24 inches. Collection of deep sediment from beneath a shallow aqueous layer can be accomplished with a system consisting of a bucket auger, a tube auger, a series of extensions and a "T" handle (Figure 1, Appendix A). The use of additional extensions can increase the depth from which sediment can be collected from 24 inches to 5 feet or more. However, water clarity must be high enough to permit the sampler to directly observe the sampling operation. In addition, sample handling and manipulation increases in difficulty with increasing depth of water. The bucket auger is used to bore a hole to the upper range of the desired sampling depth and then withdrawn. The tube auger is then lowered down the borehole, and driven into the sediment to the lower range of the desired sampling depth. The tube is then withdrawn and the sample recovered from the tube. This method can be used to collect firmly consolidated sediments, but is somewhat limited by the depth of the aqueous layer, and the integrity of the initial borehole.

The following procedure will be used to collect deep sediment samples with a bucket auger and a tube auger:

1. Attach the bucket auger to the required lengths of extensions, then attach the "T" handle to the upper extension.

2. If possible, clear the area to be sampled of any rocks or surface debris.

3. Begin augering, periodically removing any accumulated sediment (i.e., cuttings) from the auger bucket. Cuttings should be disposed of far enough from the sampling area to minimize cross contamination of various depths.

4. After reaching the upper range of the desired depth, slowly and carefully remove bucket auger from the boring.

5. Attach the tube auger to the required lengths of extensions, then attach the "T" handle to the upper extension.

6. Carefully lower the tube auger down the borehole using care to avoid making contact with the borehole sides, and cross-contaminating the sample. Gradually force the tube auger into the sediment, to the desired sampling depth. Hammering of the tube auger to facilitate coring should be avoided as the vibrations may cause the boring walls to collapse.

7. Remove the tube auger from the borehole, again taking care to avoid making contact with the borehole sides and cross contaminating the sample.

8. Discard the top of core (approximately 1 inch); this represents material collected by the tube auger before penetration to the layer of concern.

9. Transfer the sediment into an appropriate sample or homogenization container. Ensure that non-dedicated containers have been properly decontaminated.

7.2.4 Sampling Surface Sediment with an Ekman or Ponar Dredge from Beneath a Shallow Aqueous Layer or in Deep Water

For the purpose of this procedure, surface sediment is considered to range from 0 to 6 inches in depth. Collection of surface sediment can be accomplished with a system consisting of a remotely activated device (dredge) and a deployment system. This technique consists of lowering a sampling device (dredge) to the surface of the sediment by use of a rope, cable, or extended handle. The mechanism is activated, and the device entraps sediment in spring loaded or lever operated jaws.

An Ekman dredge is a lightweight sediment sampling device with spring activated jaws. It is used to collect moderately consolidated, fine textured sediment. The following procedure will be used for collecting sediment with an Ekman dredge (Figure 2, Appendix A):

1. Attach a sturdy nylon rope or stainless steel cable through the hole on the top of the bracket, or secure the extension handle to the bracket with machine bolts.

2. Fix the jaws so that they are in the open position by placing trip cables over the release studs. Ensure that the hinged doors on the dredge top are free to open.

3. Lower the sampler to a point 4 to 6 inches above the sediment surface.

4. Drop the sampler to the sediment.

5. Trigger the jaw release mechanism by lowering the messenger weight down the line, or by depressing the button on the upper end of the extension handle.

6. Raise the sampler and slowly decant any free liquid through the top of the sampler. Care should be taken to retain the fine sediment fraction during this procedure.

7. Open the dredge jaws and transfer the sediment into an appropriate container. Ensure that non-dedicated containers have been properly decontaminated.

A Ponar dredge is a heavyweight sediment sampling device with weighted jaws that are lever activated. It is used to collect consolidated fine to coarse textured sediment. The following procedure will be used for collecting sediment with a Ponar dredge (Figure 3, Appendix A):

1. Attach a sturdy nylon rope or steel cable to the ring provided on top of the dredge.

2. Arrange the Ponar dredge with the jaws in the open position, setting the trip bar so the sampler remains open when lifted from the top. If the dredge is so equipped, place the spring loaded pin into the aligned holes in the trip bar.

3. Slowly lower the sampler to a point approximately 2 inches above the sediment.

4. Drop the sampler to the sediment. Slack on the line will release the trip bar or spring loaded pin; pull up sharply on the line closing the dredge.

5. Raise the dredge to the surface and slowly decant any free liquid through the screens on top of the dredge. Care should be taken to retain the fine sediment fraction during this operation.

6. Open the dredge and transfer the sediment to an appropriate container. Ensure that non-dedicated containers have been properly decontaminated.

7.2.5 Sampling Subsurface Sediment with a Coring Device from Beneath a Shallow Aqueous Layer

For purposes of this procedure, subsurface sediment is considered to range from 6 to 18 inches in depth and a shallow aqueous layer is considered to range from 0 to 24 inches in depth. Collection of subsurface sediment from beneath a shallow aqueous layer can be accomplished with a system consisting of a tube sampler, acetate sleeve, eggshell check valve, nosecone, extensions, and "T" handle or drivehead. The use of additional extensions can increase the depth of water from which sediment can be collected from 24 inches to 10 feet or more. This sampler may be used with either a drive hammer for firm sediment, or a "T" handle for soft sediment. However, sample handling and manipulation increases in difficulty with increasing depth of water.

The following procedure describes the use of a sample coring device (Figure 4, Appendix A) used to collect subsurface sediments.

1. Assemble the coring device by inserting the acetate sleeve into the sampling tube.

2. Insert the "egg-shell" check valve into the lower end of the sampling tube with the convex surface positioned inside the acetate sleeve.

3. Screw the nosecone onto the lower end of the sampling tube, securing the acetate sleeve and egg-shell check valve. Screw the bracket to the top of the sampling tube.

4. Attach the sampling device to the required length of extensions; then attach the "T" handle or the drive hammer onto the upper extension.

5. Place the sampler in a perpendicular position on the sediment to be sampled.

6. If the "T" handle is used, place downward pressure on the device until the desired depth is reached. After the desired depth is reached, slowly withdraw the sampler from the sediment and proceed to Step 10.

7. If the drive hammer is selected, drive the sampler into the sediment to the desired depth.

8. Record the length of the tube that penetrated the sediment, and the number of blows required to obtain this depth.

9. Sharply pull the drive hammer upwards and dislodge the sampler from the sediment. Slowly withdraw the sampler from the sediment.

10. Carefully remove the coring device from the water.

11. Unscrew the nosecone and remove the eggshell check valve.

12 Slide the acetate sleeve out of the sampler tube. Decant surface water, using care to retain the fine sediment fraction. The sample may be used in this fashion, or the contents transferred to a sample or homogenization container.

13. If head space is present in the upper end, a hacksaw may be used to shear the acetate tube off at the sediment surface. The acetate core may then be capped at both ends. Indicate on the acetate tube the appropriate orientation of the sediment core using a waterproof marker.

14. The sediment may be extracted from the acetate sleeve and manipulated in the typical fashion. Extrude the sample from or open the acetate tube and transfer the sediment to an appropriate homogenization or sample container. Ensure that non-dedicated containers have been adequately decontaminated.

7.2.6 VibraCore

Sampling with a vibratory corer is divided into four steps: intrusion, extraction, core sampling, and packaging. The following procedure describes the use of a VibraCore to collect subsurface sediments.

7.2.6.1 Intrusion

The vibrator head should be attached near the top of the unsharpened end of the core barrel prior to initiating the coring procedure. After a coring location has been determined, the core pipe will be vertically positioned. The core barrel will initially sink into the sediment by its own weight, giving the barrel stability. Once the vibrator head engine is started, the pipe will rapidly penetrate into the sediment. Tying a teather line (rope) to the core barrel and pulling down by adding weight will aid in getting the pipe through resistant subsurfaces.

7.2.6.2 Extraction

After removing the vibrator head, the remaining pipe is cut off with a hacksaw approximately 2 feet above the ground surface. The distance to the sediment surface inside and outside of the pipe is measured to determine the amount of compaction. The pipe is then filled with water and a gas-main sealer plug is inserted and tightened to prevent loss of sediment from

the core pipe when it is removed.

A tripod is assembled and placed over the intruded pipe. Two come-alongs are fastened to the eyeballs on the tripod head and to a rope securely fastened to the core pipe. The core is guided through the core pipe slot in the tripod head and then rested against the tripod head to prevent falling over during extraction. When the core is completely out of the sediment, the come-alongs are removed and the core pipe slot is opened by pulling on the cord that moves the spring-loaded slot gate. The core barrel is gently placed horizontally, to prevent disturbance of the core, and examined.

7.2.6.3 Core Sampling

Sediment samples can be removed from the core either by splitting the core lengthwise and removing the sample or by drilling holes in the core liner. Splitting the core lengthwise is preferred since it allows direct observation of the sediment structure, bedding, lithologies and other features. Samples can be collected from one half of the core and the other half can be preserved for future studies or sampling. Alternatively, a power drill fitted with a 1.5- to 2-inch saw can be used to make holes in the liner. Samples can then be removed with a spoon and the hole closed by replacing the cutout disk and sealing with duct or plastic electrical tape. Spacing of approximately 1 foot is recommended to ensure that the samples are representative of the lithologies in the cores.

7.2.6.4 Packaging

If the core is to be homogenized at the laboratory, the extracted core is cut in the field using a hacksaw. Aluminum foil, plastic caps, or wooden plugs held securely with duct tape may be used to cap the core liner. Each core section must be carefully labeled, indicating the top and bottom, with a waterproof marker.

7.2.7 Diver-Assisted Core Sampling (using S.C.U.B.A. or surface-supplied air)

For the purposes of this procedure, surface sediment is considered to range from 6 to 72 inches in depth and the overhead water column is between 4 and 120 feet. Collection can be accomplished by the diver using an acetate sleeve cut to the desired sampling depth, two plastic end caps, and a metal cap and hammer. The diver may either push the core to the desired depth in soft sediment or use the metal cap and hammer to drive the core into firmer sediment. This method can be applicable in chemically and biologically hazardous environments, if the divers are properly trained, equipped, and following appropriate precautions.

The following procedure describes the use of a diver-assisted core sampling device to collect

subsurface sediments.

1. The diver is supplied with one acetate sampling tube, two plastic end caps, duct tape, a hammer, and metal hammering cap that fits over one end of the tube to receive the hammer blows. The hammering should not damage the acetate sleeve.

2. Once the sampling location is reached, the diver notes the time, depth, and any other conditions to be transferred into the appropriate logbook or sample data sheet. If on surface-supplied air, the diver communicates this information directly to the surface control.

3. The sleeve is inserted vertically into the soft sediment until the desired depth is reached. If the desired depth cannot be achieved, a metal hammer cap is assembled on top of the vertical sleeve. The diver delivers blows to the cap with a hammer until the sleeve reaches the desired depth.

4. The hammer cap is removed without disturbing the sleeve that remains at the desired depth. One plastic end cap is placed over the exposed end of the sleeve, and when possible, duct taped to secure the cap to the sleeve.

5. With the single end cap firmly in place, the sleeve is slowly removed from the sediment. In firmer sediments, a twisting or rotating motion is used to extract the sleeve.

6. While maintaining the tube vertically, a second end cap will be placed over the other end of the core to minimize any loss of material from the sleeve. Again, when possible, the cap is duct taped to secure the cap to the sleeve.

7. With both caps in place, the core is transported vertically to the surface. The diver will place their hands over the "bottom" end of the core to secure the sleeve.

8. The core will be transferred to surface personnel to maintain custody.

9. The acetate sleeve may be cut with a hacksaw at the sediment surface if headspace is present in the core. It is then recapped for shipping and storage. The sample location must be marked on each tube. The sample may be used as is, or the contents homogenized and transferred to another container.

8.0 CALCULATIONS

This section is not applicable to this SOP.

9.0 QUALITY ASSURANCE/QUALITY CONTROL

There are no specific quality assurance (QA) activities which apply to the implementation of these procedures. However, the following QA procedures apply:

1. All data must be documented on field data sheets or in site logbooks.

2. All instrumentation and equipment must be operated in accordance with the operating instructions supplied by the manufacturer, unless otherwise specified in the work plan. Equipment checkout activities must occur prior to sampling/operation, and must be documented.

10.0 DATA VALIDATION

This section is not applicable to this SOP.

11.0 HEALTH AND SAFETY

When working with potentially hazardous materials, follow U.S. EPA, Occupational Safety and Health Act (OSHA), and Corporate health and safety procedures.

More specifically, when sampling sediment from water bodies, physical hazards must be identified and adequate precautions must be taken to ensure the safety of the sampling team. The team member collecting the sample should not get too close to the edge of the water body, where bank failure may cause loss of balance. As a preventive measure, the person performing the sampling should be on a lifeline, and be wearing adequate protective equipment. This may include a personal flotation device (PFD), if necessary. If sampling from a vessel, appropriate protective measures including a PFD must be implemented.

12.0 REFERENCES

Mason, B.J. 1983. *Preparation of Soil Sampling Protocol: Technique and Strategies*. EPA-600/4-83-020.

Barth, D.S. and B.J. Mason. 1984. *Soil Sampling Quality Assurance User's Guide*. EPA-600/4-84-043.

U.S. Environmental Protection Agency. 1984. *Characterization of Hazardous Waste Sites - A Methods Manual, Available Sampling Methods*. 2nd Ed. Vol. II. EPA-600/4-84-076.

de Vera, E.R., B.P. Simmons, R.D. Stephen, and D.L. Storm. 1980. *Samplers and Sampling Procedures for Hazardous Waste Streams*. EPA-600/2-80-018.

13.0 APPENDICES

A - Figures

APPENDIX A
Figures
SOP #2016
November 2001

FIGURE 1. Sampling Auger

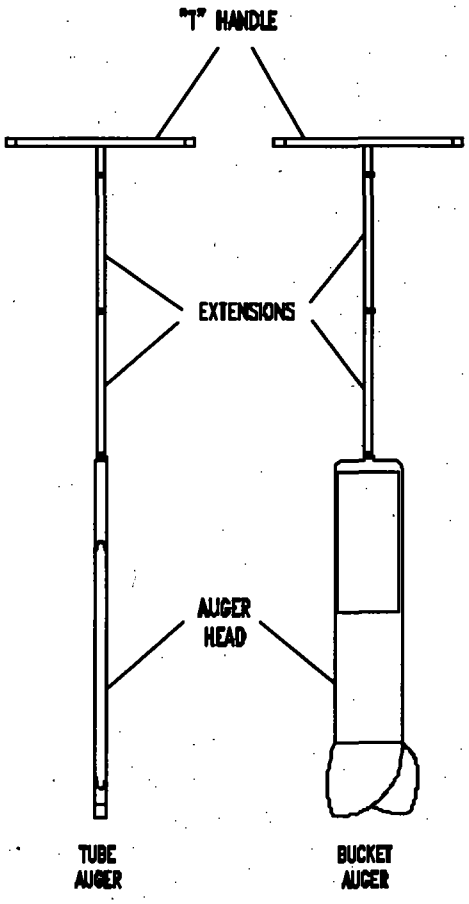


FIGURE 2. Ekman Dredge

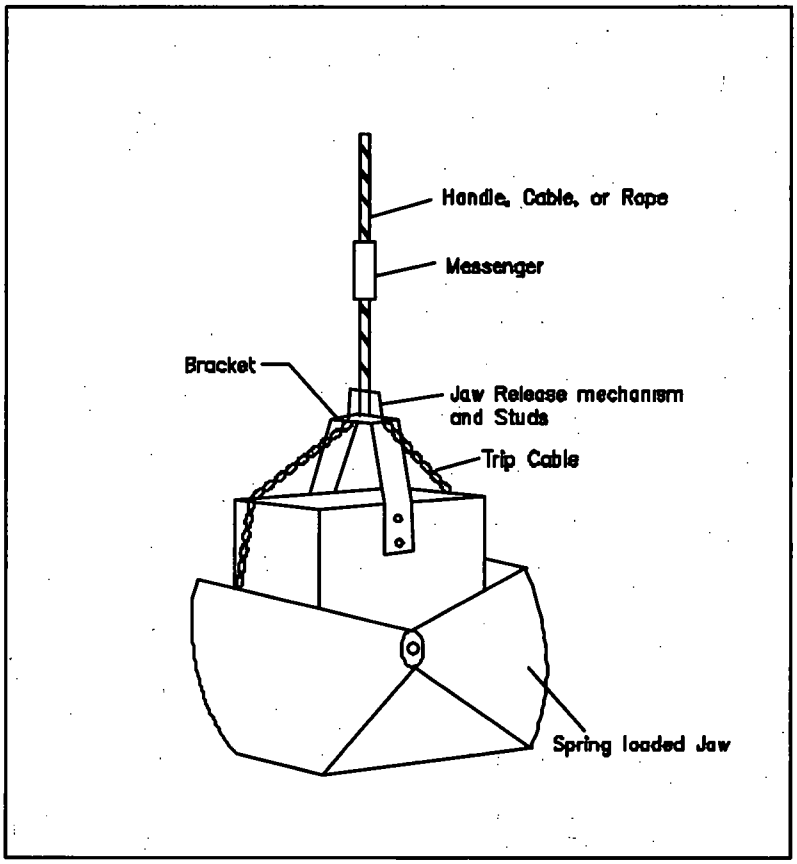


FIGURE 3. Ponar Dredge

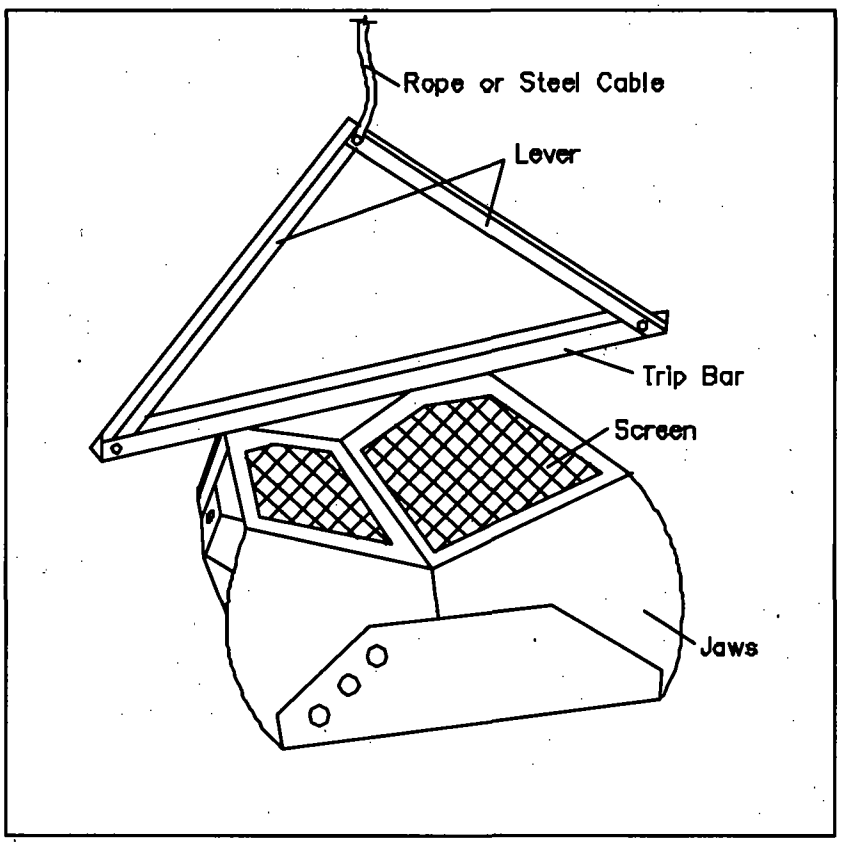
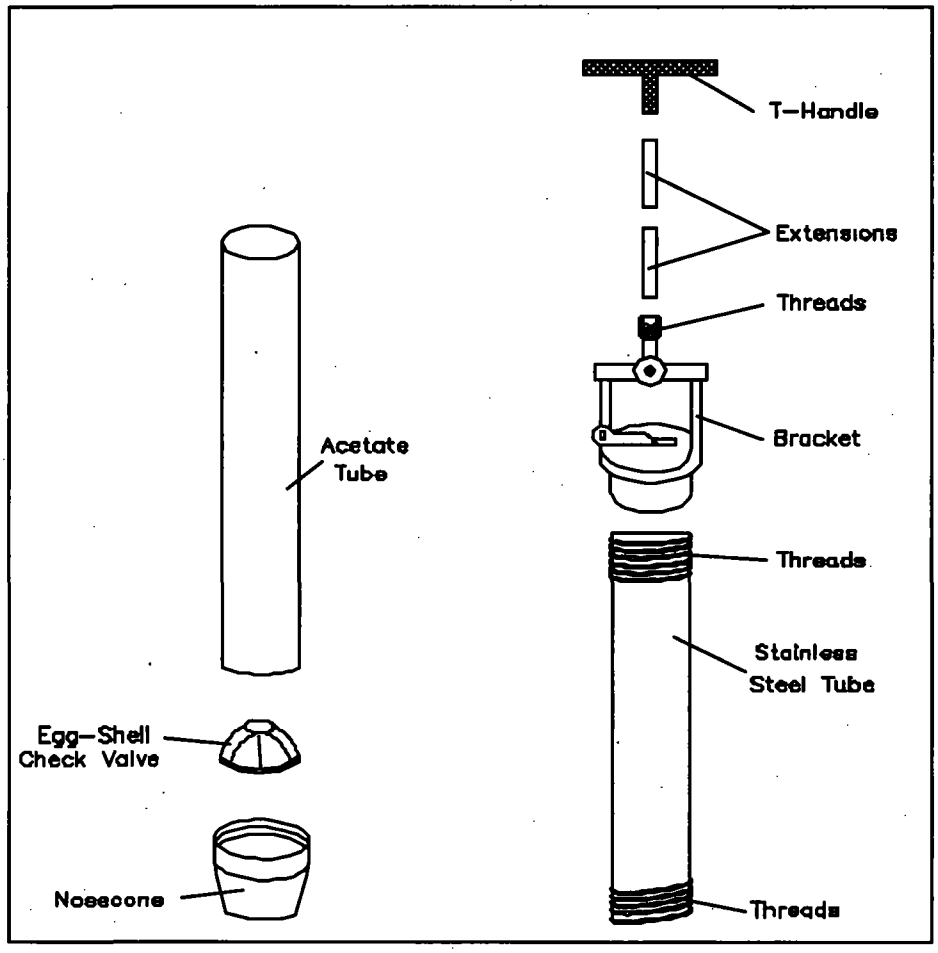

Rope or Steel Cable
Lever
Trip Bar
Screen
Jaws

FIGURE 4. Sampling Coring Device



SOIL SAMPLING

CONTENTS

*These sections affected by Revision 1.0.

SUPERCEDES: SOP #2012; Revision 0.0; 2/18/00; U.S. EPA Contract 68-C99-223.

1.0 SCOPE AND APPLICATION

The purpose of this Standard Operating Procedure (SOP) is to describe procedures for the collection of representative surface soil samples. Sampling depths are assumed to be those that can be reached without the use of a drill rig, direct-push technology, or other mechanized equipment (except for a back-hoe). Sample depths typically extend up to 1-foot below ground surface. Analysis of soil samples may define the extent of contamination, determine whether concentrations of specific contaminants exceed established action levels, or if the concentrations of contaminants present a risk to public health, welfare, or the environment.

These are standard (i.e., typically applicable) operating procedures which may be varied or changed as required, dependent upon site conditions, equipment limitations, or limitations imposed by the procedure. In all instances, the ultimate procedures employed should be documented and associated with a final report.

Mention of trade names or commercial products does not constitute United States Environmental Protection Agency (U.S. EPA) endorsement or recommendation for use.

2.0 METHOD SUMMARY

Surface soil samples can be used to investigate contaminants that are persistent in the near surface environment. Contaminants that are detected in the near surface environment may extend to considerable depths, may migrate to the groundwater, surface water, the atmosphere, or may enter biological systems.

Soil samples may be collected using a variety of methods and equipment depending on the depth of the desired sample, the type of sample required (discrete or composite), and the soil type. Near-surface soils may be easily sampled using a spade, trowel, and/or scoop. Sampling at greater depths may be performed using a hand auger, continuous-flight auger, trier, split-spoon sampler, or, if required, a backhoe.

3.0 SAMPLE PRESERVATION, CONTAINERS, HANDLING, AND STORAGE

Samples must be cooled and maintained at 4°C and protected from sunlight immediately upon collection to minimize any potential reaction. The amount of sample to be collected, proper sample container type and handling requirements are discussed in the Response Engineering and Analytical Contract (REAC) SOP #2003, *Sample Storage, Preservation and Handling.*

4.0 INTERFERENCES AND POTENTIAL PROBLEMS

There are two primary problems associated with soil sampling: 1) cross contamination of samples, and 2) improper sample collection. Cross contamination problems can be eliminated or minimized through the use of dedicated sampling equipment. If this is not possible or practical, decontamination of sampling equipment is

necessary. The guidelines for preventing, minimizing and limiting cross contamination of samples are discussed in the Environmental Response Team (ERT)/REAC SOP #2006, *Sampling Equipment Decontamination.* Improper sample collection procedures can disturb the sample matrix, resulting in volatilization of contaminants, compaction of the sample, or inadequate homogenization of the samples (when required), resulting in variable, non-representative results.

5.0 EQUIPMENT/APPARATUS

Soil sampling equipment includes the following:

- Site maps/plot plan
- Safety equipment, as specified in the site-specific Health and Safety Plan (HASP)
- Traditional survey equipment or global positioning system (GPS)
- Tape measure
- Survey stakes or flags
- Camera and image collection media
- Stainless steel, plastic*, or other appropriate homogenization bucket, bowl or pan
- Appropriate size sample containers
- Ziplock plastic bags
- Site logbook
- Labels
- Chain of Custody records and custody seals
- Field data sheets and sample labels
- Cooler(s)
- Ice
- Vermiculite
- Decontamination supplies/equipment
- Plastic sheeting
- Spade or shovel
- Spatula(s)
- Scoop(s)
- Plastic* or stainless steel spoons
- Trowel(s)
- Continuous flight (screw) auger
- Bucket auger
- Post hole auger
- Extension rods
- T-handle
- Sampling trier
- Thin wall tube sampler
- Split spoon sampler

- Soil core sampler
  - Tubes, points, drive head, drop hammer, puller jack and grip
- Photoionization detector (PID), Flame ionization detector (FID) and/or Respirable Aerosol Monitor (RAM)
- Backhoe (as required)
- En Core® samplers

* Not used when sampling for semivolatile compounds.

6.0 REAGENTS

Decontamination solutions are specified in ERT/REAC SOP #2006, *Sampling Equipment Decontamination*, and the site specific work plan.

7.0 PROCEDURES

7.1 Preparation

1. Determine the extent of the sampling effort, the analytes to be determined, the sampling methods to be employed, and the types and amounts of equipment and supplies required to accomplish the assignment.
2. Obtain the necessary sampling and air monitoring equipment.
3. Prepare schedules and coordinate with staff, client, and regulatory agencies, as appropriate.
4. Perform a general site reconnaissance survey prior to site entry in accordance with the site specific HASP.
5. Use stakes or flags to identify and mark all sampling locations. Specific site factors, including extent and nature of contamination, should be considered when selecting sample locations. If required, the proposed locations may be adjusted based on site access, property boundaries, and surface obstructions. All staked locations should be utility-cleared prior to soil sampling; utility clearances must be confirmed before beginning intrusive work.
6. Pre-clean and decontaminate equipment in accordance with the site specific work plan, and ensure that it is in working order.

7.2 Sample Collection

7.2.1 Surface Soil Samples

The collection of samples from near-surface soil can be accomplished with tools such as spades, shovels, trowels, and scoops. The over-burden or over-lying surface material is removed to the required depth and a stainless steel or plastic scoop is used to collect the sample. Plastic utensils are not to be used when sampling for semivolatile compounds.

This method can be used in most soil types but is limited to sampling at or near the ground surface. Accurate, representative samples can be collected by this procedure depending on the care and precision demonstrated by the sample team member. A flat, pointed mason trowel to cut a block of the desired soil is helpful when undisturbed profiles are required. Tools plated with chrome or other materials must not be used.

The following procedure is used to collect surface soil samples:

1. If volatile organic compound (VOC) contamination is suspected, use a PID to monitor the sampler's breathing zone during soil sampling activities.

2. Using a pre-cleaned, stainless steel scoop, plastic spoon, or trowel, remove and discard sticks, rocks, vegetation and other debris from the sampling area.

3. Accumulate an adequate volume of soil, based on the type(s) of analyses to be performed, in a stainless, plastic or other appropriate container.

4. If volatile organic analysis is to be performed, immediately transfer the sample directly into an appropriate, labeled sample container with a stainless steel spoon, or equivalent, and secure the cap tightly to ensure that the volatile fraction is not compromised. Thoroughly mix the remainder of the soil to obtain a sample that is representative of the entire sampling interval. Then, either place the sample into appropriate, labeled containers and secure the caps tightly, or, if composite samples are to be collected, place a sample from another sampling interval or location into the homogenization container and mix thoroughly. When compositing is complete, place the sample into appropriate, labeled containers and secure the caps tightly.

7.2.2 Sampling at Depth with Augers and Thin Wall Tube Samplers

This system consists of an auger, head, a series of extensions, and a "T" handle (Figure 1, Appendix A). The auger is used to bore a hole to a desired sampling depth, and is then withdrawn. The sample may be collected directly from the auger head. If additional sample volume is required, multiple grabs at the same depth are made. If a core sample

is to be collected, the auger head is then replaced with a tube auger. The system is then lowered down the borehole, and driven into the soil to the completion depth. The system is withdrawn and the core is collected.

Several types of augers are available; these include bucket or tube type, and continuous flight (screw) or post-hole augers. Bucket or tube type augers are better for direct sample recovery because a large volume of sample can be collected from a discrete area in a short period of time. When continuous flight or post-hole augers are used, the sample can be collected directly from the flights or from the borehole cuttings. The continuous flight or post-hole augers are satisfactory when a composite of the complete soil column is desired, but have limited utility for sample collection as they cannot be used to sample a discrete depth.

The following procedure is used for collecting soil samples with an auger:

1. Attach the auger head to an extension rod and attach the "T" handle.

2. Clear the area to be sampled of surface debris (e.g., twigs, rocks, litter). It may be advisable to remove a thin layer of surface soil for an area approximately six inches in radius around the sampling location.

3. Begin augering, periodically removing and depositing accumulated soils onto a plastic sheet spread near the hole. This prevents the accidental brushing of loose material back down the borehole when removing the auger or adding extension rods. It also facilitates refilling the hole, and avoids possible contamination of the surrounding area.

4. After reaching the desired depth, slowly and carefully remove the auger from the hole. When sampling directly from the auger head, proceed to Step 10.

5. Remove auger tip from the extension rods and replace with a tube sampler. Install the proper cutting tip.

6. Carefully lower the tube sampler down the borehole. Gradually force the tube sampler into the soil. Do not scrape the borehole sides. Avoid hammering the rods as the vibrations may cause the boring walls to collapse.

7. Remove the tube sampler and unscrew the extension rods.

8. Remove the cutting tip and the core from the device.

9. Discard the top of the core (approximately 1 inch), as this possibly represents material collected before penetration of the layer of concern. Place the core or a discrete portion of the core into the appropriate labeled sample container using a clean, decontaminated stainless steel spoon. If required, homogenize the sample as described in Step 10.

10. If VOC analysis is to be performed, transfer the sample directly from the auger head into an appropriate, labeled sample container with a stainless steel spoon, or equivalent and secure the cap tightly.

11. If another sample is to be collected in the same hole, but at a greater depth, reattach the auger head to the drill assembly, and follow steps 3 through 11, making sure to decontaminate the auger head and tube sampler between samples.

12. Abandon the hole according to applicable state regulations.

7.2.3 Sampling at Depth with a Trier

The system consists of a trier and a "T" handle. The auger is driven into the soil to be sampled and used to extract a core sample from the appropriate depth.

The following procedure is used to collect soil samples with a sampling trier:

1. Insert the trier (Figure 2, Appendix A) into the material to be sampled at a zero degree to forty-five degree (0° to 45°) angle from the soil surface plane. This orientation minimizes the spillage of sample.

2. Rotate the trier once or twice to cut a core of material.

3. Slowly withdraw the trier, making sure that the slot is facing upward.

4. If VOC analyses are required, transfer the sample directly from the trier into an appropriate, labeled sample container with a stainless steel spoon, or equivalent device and secure the cap tightly. Place the remainder of the sample into a stainless steel, plastic, or other appropriate homogenization container and mix thoroughly to obtain a sample that is representative of the entire sampling interval. Then, either place the sample into appropriate, labeled containers and secure the caps tightly; if composite samples are to be collected, place a sample from another sampling interval into the homogenization container and mix thoroughly. When compositing is complete, place the sample into appropriate, labeled containers and secure the caps tightly.

7.2.4 Sampling at Depth with a Split Spoon (Barrel) Sampler

Split spoon sampling is generally used to collect undisturbed soil cores of 18- or 24-inches in length. A series of consecutive cores may be extracted with a split spoon sampler to give a complete soil column profile, or an auger may be used to drill down to the desired depth for sampling. The split spoon is then driven to its sampling depth through the bottom of the augured hole and the core extracted.

When split spoon sampling is performed to gain geologic information, all work should be performed in accordance with American Society for Testing and Materials (ASTM) D1586-99, "*Standard Test Method for Penetration Test and Split-Barrel Sampling of Soils*".

The following procedures are used for collecting soil samples with a split spoon:

1. Assemble the sampler by aligning both sides of the barrel and then screwing the drive shoe on the bottom and the head piece on top.

2. Place the sampler at a 90 degree (90°) angle to the sample material.

3. Using a well ring, drive the sampler. Do not drive past the bottom of the head piece or compression of the sample will result.

4. Record in the site logbook or on field data sheets the length of the tube used to penetrate the material being sampled, and the number of blows required to obtain the sample.

5. Withdraw the sampler, and open it by unscrewing the bit and head, and then splitting the barrel. The amount of recovery and soil type should be recorded on the boring log. If a split sample is desired, a cleaned, stainless steel knife should be used to divide the tube contents in half, longitudinally. This sampler is typically available in 2- and 3.5-inch diameter tubes. A larger barrel (diameter and/or length) may be necessary to obtain the required sample volume.

6. Without disturbing the core, transfer it to the appropriately labeled sample container(s) and seal tightly. Place the remainder of the sample into a stainless steel, plastic, or appropriate homogenization container, and mix thoroughly to obtain a sample that is representative of the entire sampling interval. Then, either place the sample into the appropriate, labeled containers and secure the caps tightly, or if composite samples are to be collected, place a sample from another sampling interval or location into the homogenization container and mix

thoroughly. When compositing is complete, place the sample into the appropriate, labeled containers and secure the caps tightly.

7. Abandon the hole according to applicable state regulations.

7.2.5 Test Pit/Trench Excavation

A backhoe can be used to remove sections of soil when a detailed examination of stratigraphy and soil characteristics is required. The following procedures are used for collecting soil samples from test pits or trenches:

1. Prior to any excavation with a backhoe, it is imperative to ensure that all sampling locations are clear of overhead and buried utilities.

2. Review the site specific HASP and ensure that all safety precautions including appropriate monitoring equipment are installed as required.

3. Using the backhoe, excavate a trench approximately three feet wide and approximately one foot deep below the cleared sampling location. Place excavated soils on plastic sheets. Trenches greater than five feet deep must be sloped or protected by a shoring system, as required by Occupational Safety and Health Administration (OSHA) regulations.

4. A shovel is used to remove a one to two inch layer of soil from the vertical face of the pit where sampling is to be done.

5. Samples are taken using a trowel, scoop, or coring device at the desired intervals. Be sure to scrape the vertical face at the point of sampling to remove any soil that may have fallen from above, and to expose fresh soil for sampling. In many instances, samples can be collected directly from the backhoe bucket.

6. If VOC analyses are required, transfer the sample into an appropriate, labeled sample container with a stainless steel spoon, or equivalent and secure the cap tightly. Place the remainder of the sample into a stainless steel, plastic, or other appropriate homogenization container, and mix thoroughly to obtain a sample representative of the entire sampling interval. Then, either place the sample into appropriate, labeled containers and secure the caps tightly; or, if composite samples are to be collected, place a sample from another sampling interval into the homogenization container and mix thoroughly. When compositing is complete, place the sample into the appropriate, labeled containers and secure the caps tightly.

7. Abandon the pit or excavation according to applicable state regulations.

7.2.6 Sampling for VOCs in Soil Using an En Core® Sampler

An En Core® sampler is a single-use device designed to collect and transport samples to the laboratory. The En Core® sampler is made of an inert composite polymer and reduces the open-air handling of soil samples in the field and in the laboratory; thereby, minimizing losses of VOCs.

1. Assemble the coring body, plunger rod and T-handle according to the instructions provided with the En Core® sampler.

2. Turn the T-handle with the T-up and the coring body down and push the sampler into the soil until the coring body is completely full. Remove the sampler from the soil. Wipe excess soil from the coring body exterior.

3. Cap the coring body while it is still on the T-handle. Push the cap over the flat area of the ridge. Be sure that the cap is seated properly to seal the sampler. Push and cap to lock arm in place.

4. Remove the capped sampler by depressing the locking lever on the T-handle while twisting and pulling the sampler from the T-handle.

5. Attach the label to the coring body cap, place in a plastic zippered bag, seal and put on ice.

Generally, three En Core® samplers are required for each sample location. These samplers are shipped to the laboratory where the cap is removed and the soil samples are preserved with methanol or sodium bisulfate.

8.0 CALCULATIONS

This section is not applicable to this SOP.

9.0 QUALITY ASSURANCE/QUALITY CONTROL

There are no specific quality assurance (QA) activities that apply to the implementation of these procedures. However, the following general QA procedures apply:

1. All data must be documented in site logbooks or on field data sheets. At a minimum, the following data is recorded:

Sampler's name and affiliation with project
Sample number
Sample location
Sample depth
Approximate volume of sample collected
Type of analyses to be performed
Sample description
Date and time of sample collection
Weather conditions at time of sampling
Method of sample collection
Sketch of sample location

2. All instrumentation must be operated in accordance with applicable SOPs and/or the manufacturer's operating instructions, unless otherwise specified in the work plan. Equipment checkout and calibration activities must occur prior to sampling/operation, and must be documented.

3. The types of quality control (QC) samples to be collected in the field shall be documented in the site-specific Work Plan.

10.0 DATA VALIDATION

This section is not applicable to this SOP.

11.0 HEALTH AND SAFETY

When working with potentially hazardous materials, follow U.S. EPA, OSHA and corporate health and safety procedures, in addition to the procedures specified in the site specific HASP.

12.0 REFERENCES

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Barth, D.S. and B.J. Mason. 1989. *Soil Sampling Quality Assurance User's Guide*. EPA-600/8-89-046.

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American Society for Testing and Materials. *Standard Test Method for Penetration Test and Split-Barrel Sampling of Soils*. Method D 1586-99.

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APPENDIX A
Figures
SOP #2012
July 2001

FIGURE 1. Sampling Augers

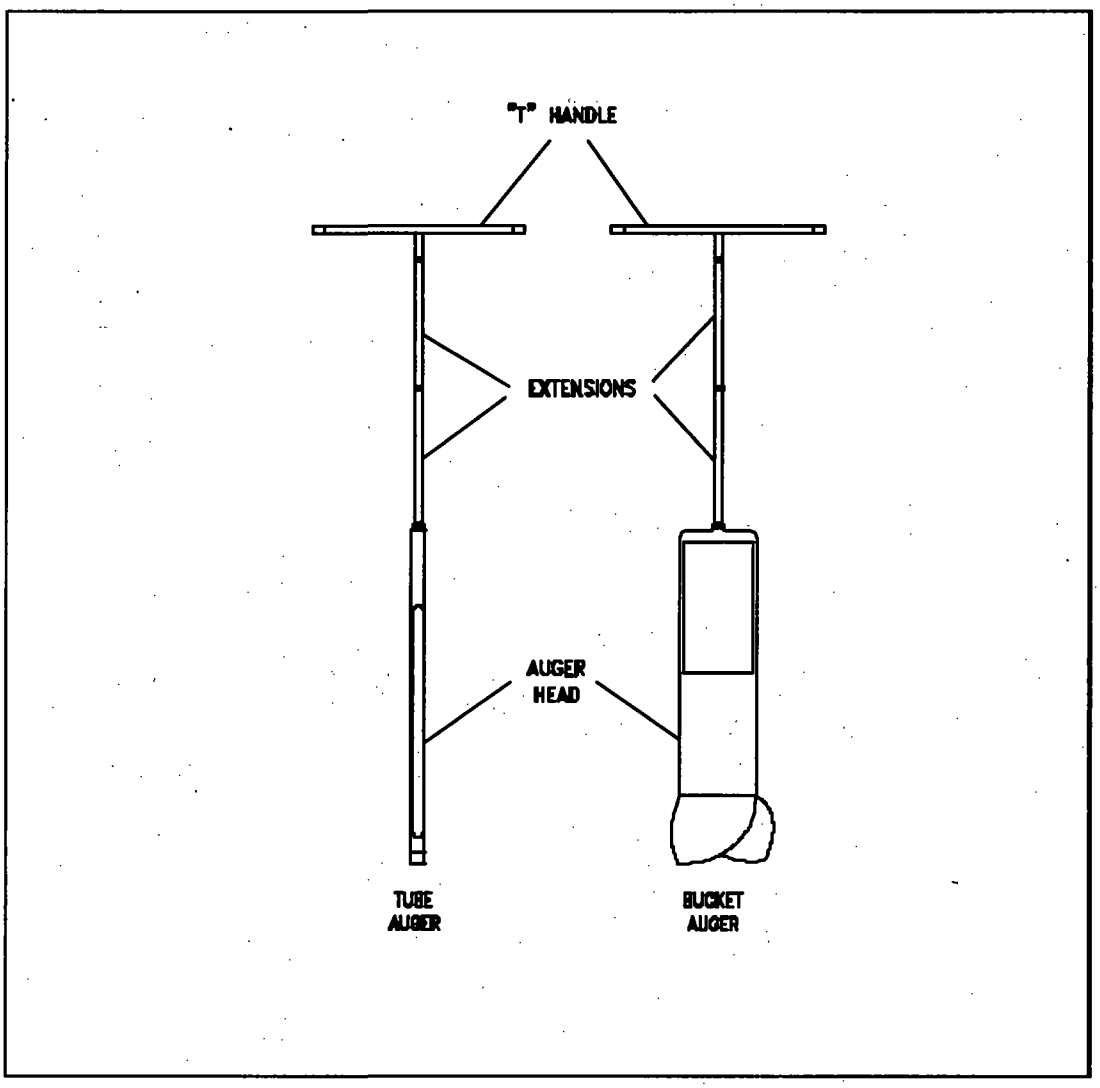
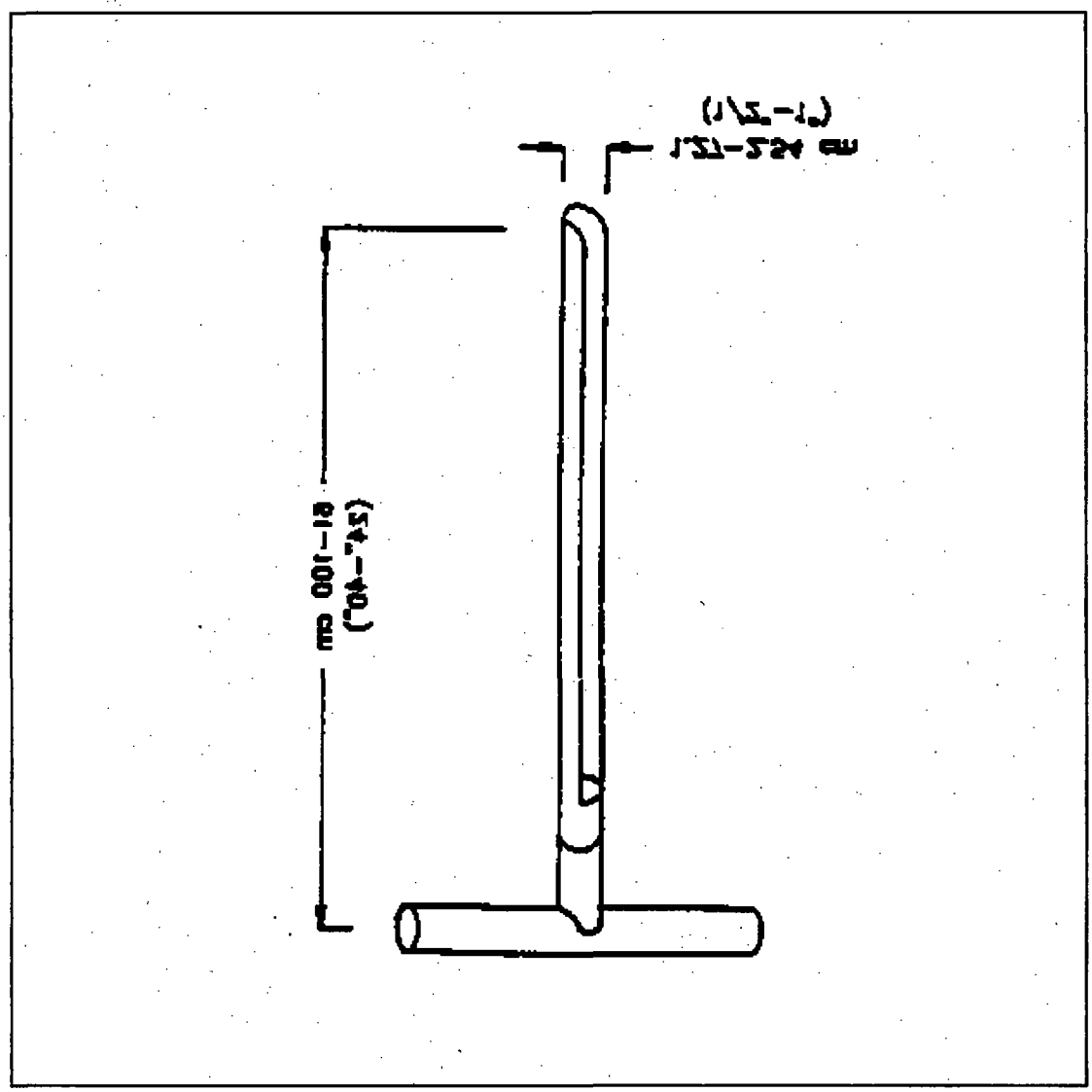


FIGURE 2. Sampling Trier



REAC
Response Engineering and Analytical Contract

STANDARD OPERATING PROCEDURES

SURFACE WATER SAMPLING

CONTENTS

CONTENTS (Cont'd)

* These sections affected by Revision 0.0.

SUPERSEDES: SOP #2013; Revision 0.0; 11/17/94; U.S. EPA Contract 68-C4-0022.

1.0 SCOPE AND APPLICATION

This standard operating procedure (SOP) is applicable to the collection of representative surface water samples from streams, rivers, lakes, ponds, lagoons, and surface impoundments. It includes samples collected from depth, as well as samples collected from the surface.

These are standard (i.e., typically applicable) operating procedures which may be varied or changed as required, dependent upon site conditions, equipment limitations or limitations imposed by the procedure. In all instances, the ultimate procedures employed should be documented and associated with the final report.

Mention of trade names or commercial products does not constitute United States Environmental Protection Agency (U.S. EPA) endorsement or recommendation for use.

2.0 METHOD SUMMARY

Sampling situations vary widely; therefore, no universal sampling procedure can be recommended. However, surface water sampling is generally accomplished through the use of one of the following samplers or techniques:

- Kemmerer bottle
- Van Doren sampler
- Bacon bomb sampler
- Dip sampler
- Direct method

These samplers and sampling techniques will result in the collection of representative samples from the majority of surface waters and impoundments encountered.

3.0 SAMPLE PRESERVATION, CONTAINERS, HANDLING, AND STORAGE

Once samples have been collected, the following procedures should be followed:

1. Transfer the sample(s) into suitable, labeled sample containers specific for the analyses to be performed.

2. Preserve the sample, if appropriate, or use pre-preserved sample bottles. Do not overfill bottles if they are pre-preserved.

3. Cap the container securely, place in a resealable plastic bag, and cool to 4°C.

4. Record all pertinent data in the site logbook and/or on field data sheets.

5. Complete the Chain of Custody record.

6. Attach custody seals to cooler prior to shipment.

7. Decontaminate all non-dedicated sampling equipment prior to the collection of additional samples.

4.0 INTERFERENCES AND POTENTIAL PROBLEMS

There are two primary interferences or potential problems associated with surface water sampling. These include cross contamination of samples and improper sample collection.

1. Cross contamination problems can be eliminated or minimized through the use of dedicated or disposable sampling equipment. If this is not possible or practical, then decontamination of sampling equipment is necessary. Refer to ERT/REAC SOP #2006, *Sampling Equipment Decontamination.*

2. Improper sample collection can involve using contaminated equipment, disturbance of the stream or impoundment substrate, and sampling in an obviously disturbed or non-representative area.

Following proper decontamination procedures, minimizing disturbance of the sample site, and careful selection of sampling locations will eliminate these problems. Proper timing for the collection of samples must be taken into consideration due to tidal influences and low or fast-flowing streams or rivers.

5.0 EQUIPMENT/APPARATUS

Equipment needed for collection of surface water samples may include (depending on technique chosen):

- Kemmerer bottles
- Van Doren sampler
- Bacon bomb sampler
- Dip sampler
- Line and messengers
- Peristalic pump
- Tygon tubing
- 0.45 micron (• m) filters
- Sample bottles/preservatives
- pH paper
- Resealable plastic bags
- Ice
- Coolers, packing material
- Chain of Custody records, custody seals
- Field data sheets

- Decontamination equipment/supplies
- Maps/plot plan
- Safety equipment
- Compass
- Tape measure
- Survey stakes, flags, or buoys and anchors
- Camera and film
- Logbook/waterproof pen
- Sample bottle labels
- Paper towels
- Disposable pipets
- Hydrolab

6.0 REAGENTS

Reagents will be utilized for preservation of samples and for decontamination of sampling equipment. The preservatives required are specified by the analysis to be performed and are summarized in ERT/REAC SOP #2003, *Sample Storage, Preservation and Handling*. Decontamination solutions are specified in ERT/REAC SOP #2006, *Sampling Equipment Decontamination.*

7.0 PROCEDURES

7.1 Preparation

1. Determine the extent of the sampling effort, the sampling methods to be employed, and the types and amounts of equipment and supplies needed.

2. Obtain the necessary sampling and monitoring equipment.

3. Decontaminate or pre-clean equipment, and ensure that it is in working order.

4. Prepare scheduling and coordinate with staff, clients, and regulatory agency, if appropriate.

5. Perform a general site survey prior to site entry, in accordance with the site specific Health and Safety Plan (HASP).

6. Use stakes, flags, or buoys to identify and mark all sampling locations. If required, the proposed locations may be adjusted based on site access, property boundaries, and obstructions.

7.2 Representative Sampling Considerations

In order to collect a representative sample, the hydrology and morphometrics of a stream, river, pond, lake or impoundment should be determined prior to sampling. This will aid in determining the presence of phases or layers in lagoons or impoundments, flow patterns in streams, and appropriate sample locations and depths.

Water quality data should be collected in ponds, lakes and impoundments to determine if stratification is present. Measurements of dissolved oxygen, pH, conductivity, oxidation-potential, temperature and turbidity can indicate if strata exist that would affect analytical results. Measurements should be collected at one-meter intervals from the surface to the bottom using the appropriate instrument (i.e., a Hydrolab or equivalent). These water quality measurements can assist in the interpretation of analytical data, and the selection of sampling sites and depths when surface water samples are collected.

Factors that contribute to the selection of a sampling device used for sampling surface waters in streams, rivers, lakes, ponds, lagoons, and surface impoundments are:

- Width, depth, flow and accessibility of the location being sampled
- Whether the sample will be collected onshore or offshore

7.2.1 Sampler Composition

The appropriate sampling device must be of a proper composition. Selection of samplers constructed of glass, stainless steel, polyvinyl chloride (PVC) or PFTE (Teflon®) should be based upon the suspected contaminants and the analyses to be performed.

7.3 Sample Collection

7.3.1 Kemmerer Bottle

A Kemmerer bottle (Figure 1, Appendix A) may be used in most situations where site access is from a boat or structure, such as a bridge or pier, and where samples at specific depths are required. Sampling procedures are as follows:

1. Use a properly decontaminated Kemmerer bottle. Set the sampling device so that the upper and lower stoppers are pulled away from the body, allowing the surface water to enter tube.

2. Lower the pre-set sampling device to the predetermined depth. Avoid disturbance of the bottom.

3. When the Kemmerer bottle is at the required depth, send the weighted messenger down the suspension line, closing the sampling device.

4. Retrieve the sampler and discharge the first 10-20 milliliters (mL) from the drain to clear potential contamination from the valve. This procedure may be repeated if additional sample volume is needed to fulfill analytical requirements. Subsequent grabs may be composited or transferred directly to appropriate sample containers.

7.3.2 Van Doren Sampler

A Van Doren sampler (Figure 2, Appendix A) is used to collect a surface water from a very specific sampling depth or from a shallow water body. Since the sampler is suspended horizontally, the depth interval sampled is the diameter of the sampling tube. The sampling procedure is as follows:

1. Use a properly decontaminated Van Doren sampler. Set the device so that the end stoppers are pulled away from the body allowing surface water to enter the tube.

2. Lower the pre-set sampling device to the predetermined depth. Avoid disturbance of the bottom.

3. When the Van Doren is at the required depth, send the weighted messenger down the suspension line, closing the sampling device.

4. Retrieve the sampler and discharge the first 10-20 milliliters (mL) from the drain to clear potential contamination from the valve. This procedure may be repeated if additional sample volume is needed to fulfill analytical requirements. Subsequent grabs may be composited or transferred directly to appropriate sample containers.

7.3.3 Bacon Bomb Sampler

A bacon bomb sampler (Figure 3, Appendix A) may be used in situations similar to those outlined for the Kemmerer bottle. Sampling procedures are as follows:

1. Lower the bacon bomb sampler carefully to the desired depth, allowing the line for the trigger to remain slack at all times. When the desired depth is reached, pull the trigger line until taut. This will allow the sampler to fill.

2. Release the trigger line and retrieve the sampler.

3. Discharge the first 10-20 milliliters (mL) from the drain to clear potential contamination from the valve. This procedure may be repeated if additional sample volume is needed to fulfill analytical requirements. Subsequent grabs may be composited or transferred directly to appropriate sample containers.

7.3.4 Dip Sampler

A dip sampler (Figure 4, Appendix A) is useful in situations where a sample is to be recovered from an outfall pipe or along a lagoon bank where direct access is limited. The long handle on such a device allows access from a discrete location. Sampling procedures are as follows:

1. Assemble the device in accordance with the manufacturer's instructions.

2. Extend the device to the sample location and collect the sample by dipping the sampler into the water.

3. Retrieve the sampler and transfer the sample to the appropriate sample container(s).

7.3.5 Direct Method

For streams, rivers, lakes, and other surface waters, the direct method may be utilized to collect water samples directly into the sample container(s). Health and safety considerations must be addressed when sampling lagoons or other impoundments where specific conditions may exist that warrant the use of additional safety equipment. These issues must be addressed in the site-specific HASP.

Using adequate protective clothing, access the sampling station by appropriate means. For shallow stream stations, collect the sample under the water surface while pointing the sample container upstream; the container must be upstream of the collector. Avoid disturbing the substrate. For lakes and other impoundments, collect the sample under the water surface while avoiding surface debris and the boat wake.

When using the direct method, do not use pre-preserved sample bottles as the collection method may dilute the concentration of preservative necessary for proper sample preservation.

8.0 CALCULATIONS

This section is not applicable to this SOP.

9.0 QUALITY ASSURANCE/QUALITY CONTROL

There are no specific quality assurance (QA) activities which apply to the implementation of these procedures. However, the following general QA procedures apply:

1. All data must be documented on field data sheets or within site logbooks.

2. All instrumentation must be operated in accordance with operating instructions as supplied by the manufacturer, unless otherwise specified in the work plan. Equipment checkout and calibration activities must occur prior to sampling/operation and they must be documented.

3. To avoid the incidental inclusion of disturbed sediment in the sample, surface water should be collected from a downstream to upstream direction and upstream of any activity that may disturb the sediment (i.e., wading).

4. While collecting surface water using the direct method, the sample container should be held below the surface to avoid the collection of floating debris.

5. Water quality data should be collected to detect the presence of stratified layers or other site-specific characteristics that would affect the sample.

10.0 DATA VALIDATION

This section is not applicable to this SOP.

11.0 HEALTH AND SAFETY

When working with potentially hazardous materials, follow U.S. EPA, Occupational Health and Safety (OSHA) and corporate health and safety procedures.

More specifically, when sampling lagoons or surface impoundments containing known or suspected hazardous substances, adequate health and safety and boating precautions must be taken to ensure the safety of sampling personnel.

12.0 REFERENCES

Wilde, F.D., D.B. Radtke, J. Gibs and R.T. Iwatsubo. 1998. National Field Manual for the Collection of Water-Quality Data - Selection of Equipment for Water Sampling. U.S. Geological Survey Techniques of Water - Resources Investigations, Book 9, Chap. A2, variously paged.

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13.0 APPENDICES

A - Figures

APPENDIX A
Figures
SOP #2013
February 2002

FIGURE 1. Kemmerer Bottle

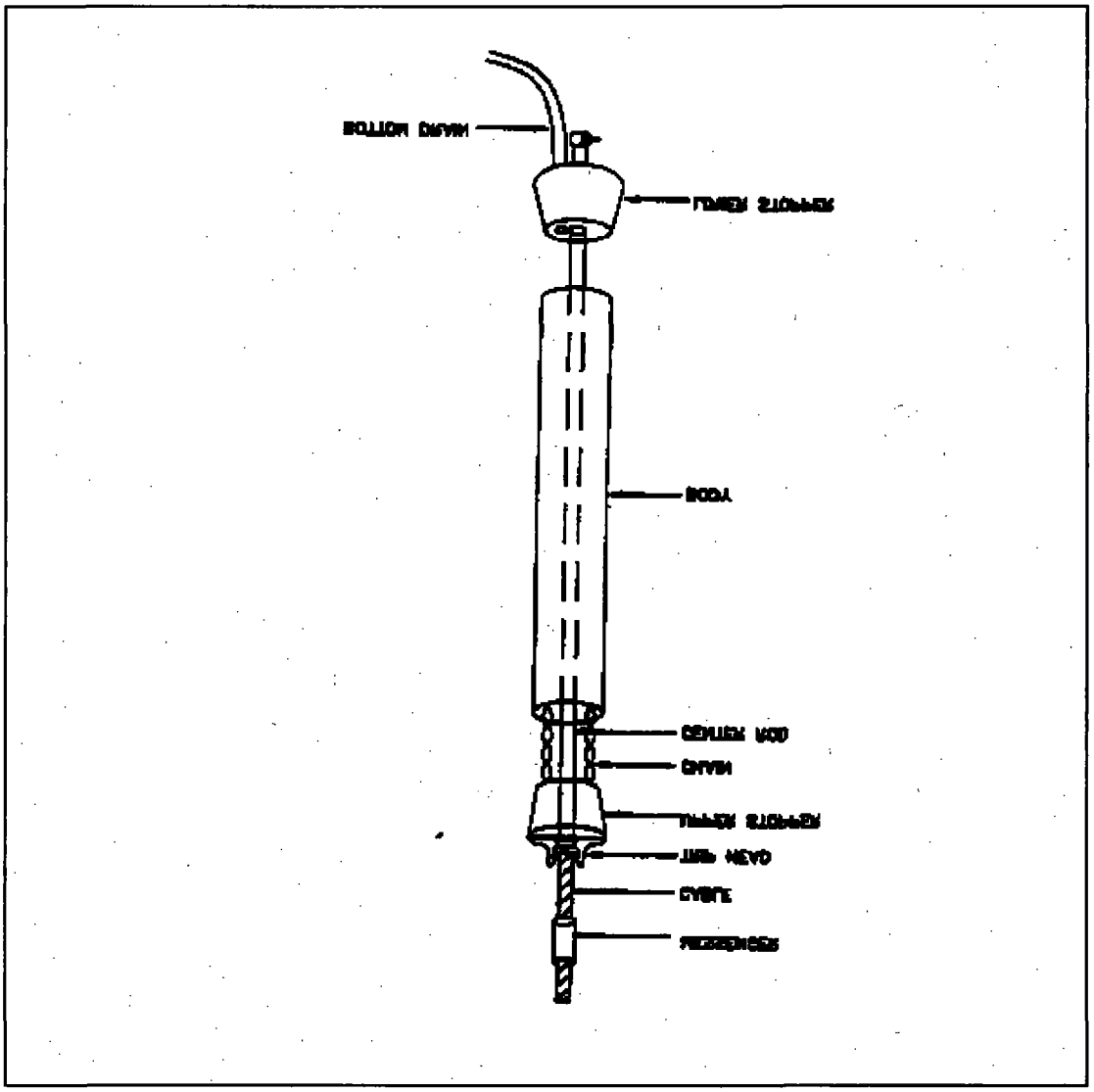

FIGURE 2. Van Doren Sampler

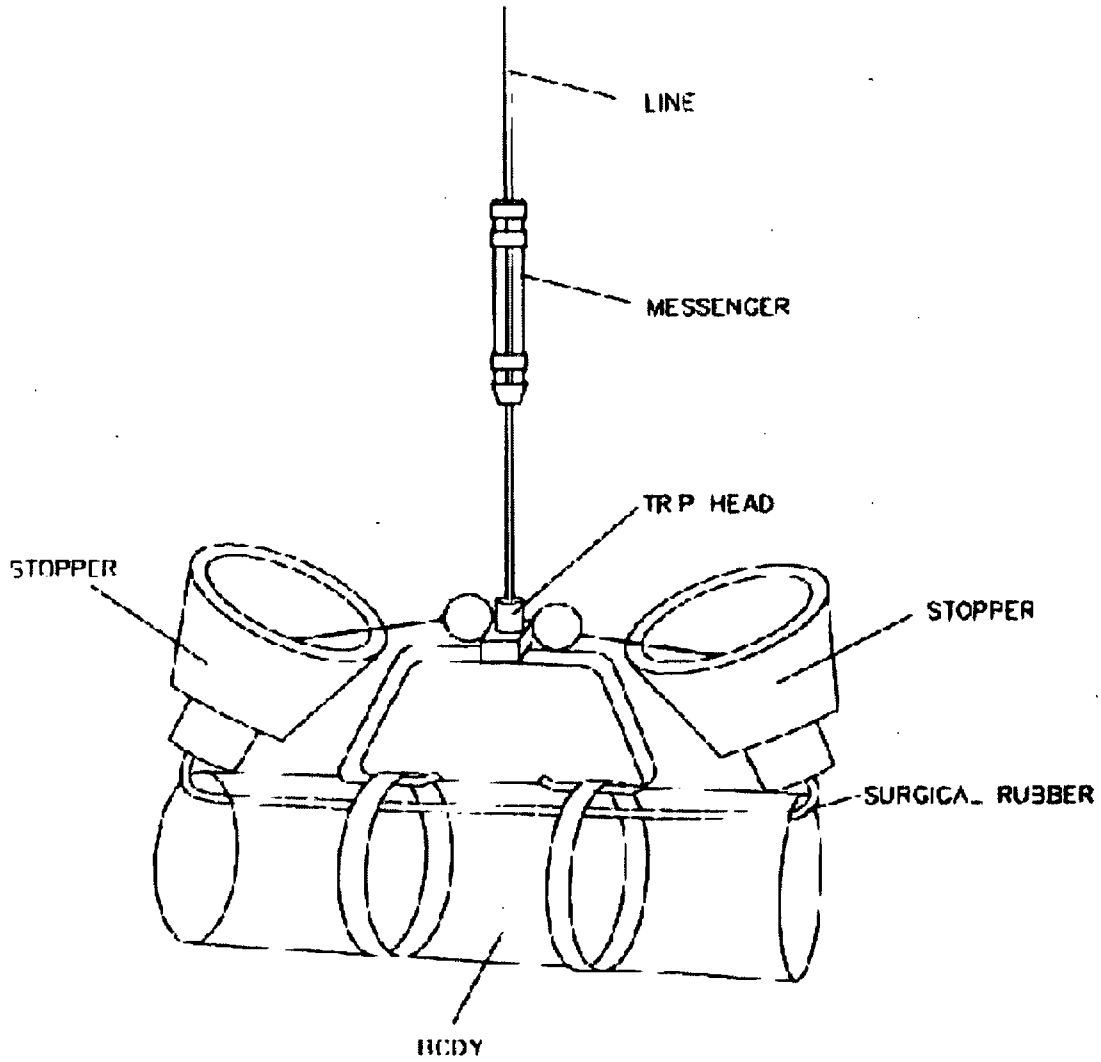

FIGURE 3. Bacon Bomb Sampler

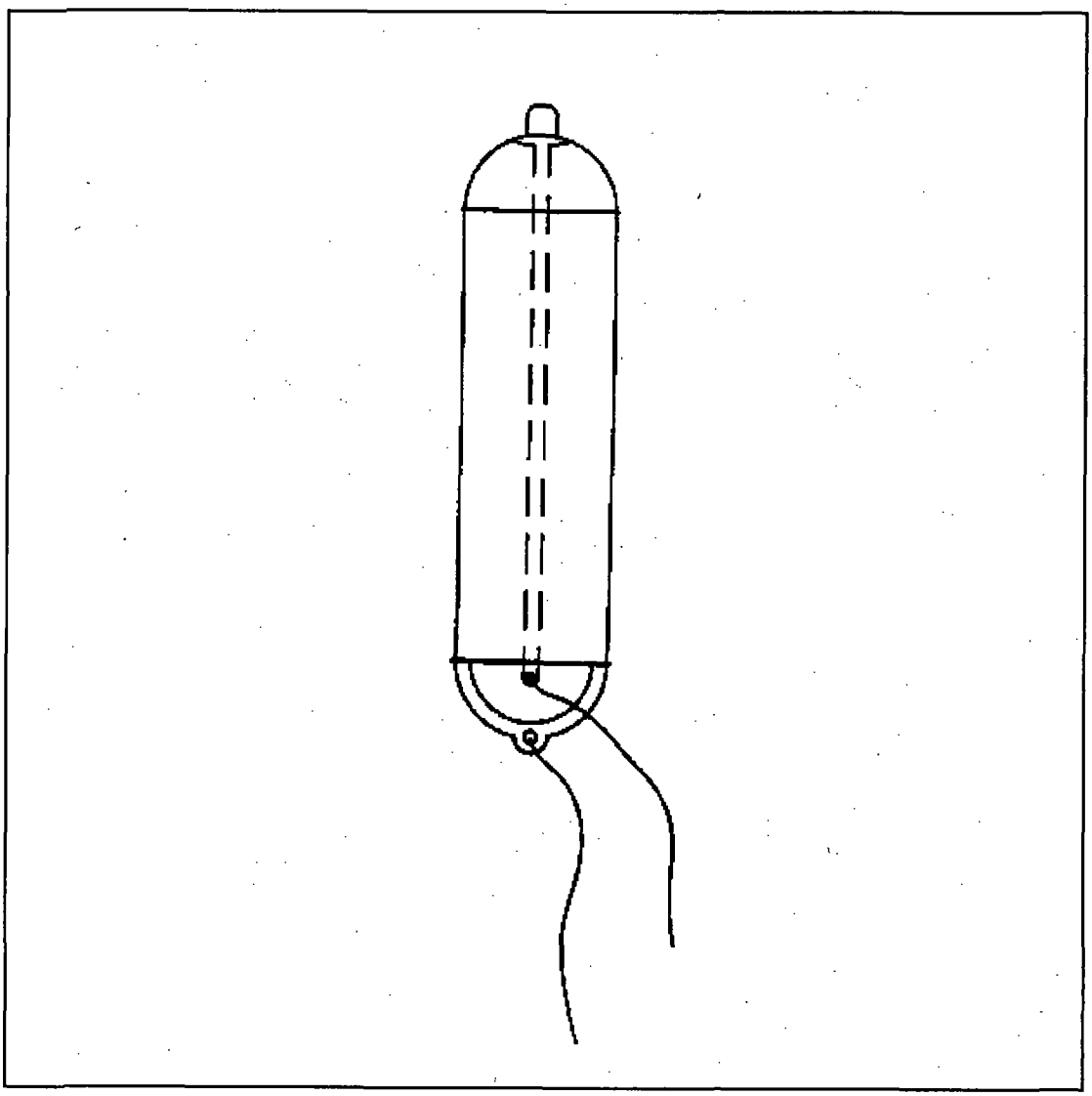

FIGURE 4. Dip Sampler